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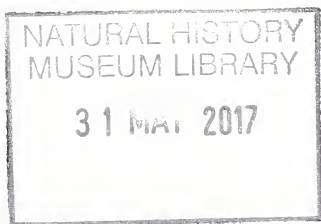


COVER: *Leptoglossus occidentalis* (eggs)

Freshly-laid eggs of *Leptoglossus occidentalis*, showing intricate surface details and the opercula through which the nymphs emerge.

Photograph details:

Photograph by Ward Strong (Ministry Forests, Lands and Natural Resource Operations). Photo taken with a Nikon D90 mounted on a Leica stereozoom microscope, stack of 43 focus-bracketed images stitched with Helicon Focus software.



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Lodgepole pine seedset increase by mesh bagging is due to exclusion of *Leptoglossus occidentalis* (Hemiptera: Coreidae).

WARD B. STRONG¹

ABSTRACT

Lodgepole pine (*Pinus contorta* Dougl. ex. Loud. var *latifolia*) seed orchards in British Columbia, Canada, suffer from low seedset problems. Seedset can be improved by installing mesh bags over the second-year cones. One mechanism of improved seedset has been shown to be exclusion of the seed-feeding western conifer seedbug, *Leptoglossus occidentalis*, but the possibility of microclimate alteration leading to improved seedset has not been carefully explored. I measured the internal cone temperatures of cones both inside and outside mesh bags, as well as the relative humidity around cones both inside and outside bags. I also installed time-lapse cameras that, every 5 minutes, photographed unbagged cones situated near bagged cones, and examined the photographs for feeding by *L. occidentalis*. Internal temperatures of cones inside bags were warmer, accumulated more heat units, and had higher peak temperatures than cones outside bags. Humidity was slightly higher inside bags in late season, particularly during rainy days. The evidence did not support the hypothesis that microclimate change is the cause of improved seedset inside bags. On the other hand, the cameras documented extensive feeding by *L. occidentalis* on unbagged cones, and showed a strong correlation between the amount of time a cone was fed upon and the subsequent seed loss. Therefore the increase in seedset due to the mesh bags is not related to microclimate change, but is caused by exclusion of *L. occidentalis*.

INTRODUCTION

Conifer seed orchards produce seed from commercial forestry tree species that have been genetically improved through traditional breeding techniques. In British Columbia (BC), Canada, seed orchards provided seeds in 2012 for 243 million seedlings used in reforestation (Forest Genetics Council of BC 2013a). However, lodgepole pine (*Pinus contorta* Dougl. ex. Loud. Var. *latifolia*) orchards have not been meeting production targets due to low seedset issues (Forest Genetics Council of BC 2013b).

Lodgepole pine seedset, as measured by the number of filled seeds per cone or percent filled seeds, has been below the potential maximum in all BC seed orchards except those at the Prince George Tree Improvement Station. The potential number of filled seeds is between 30 and 40 per cone (Owens 2006), but many orchards experience fewer than 15, even as low as 3 to 5, filled seeds per cone (Stephen Joyce², personal communication). One factor influencing seedset is the western conifer seedbug, *Leptoglossus occidentalis* (Heidemann) (Hemiptera: Coreiidae) (Hedlin *et al.* 1980; Koerber 1963), which has been shown to reduce seed crops significantly (Bates *et al.* 2000; Blatt & Borden 1996; Strong 2006).

It was discovered in 2010 that a substantial amount of seed loss occurs from late July through August; this was substantiated in 2011 and 2012 (WBS, unpublished data). Though seasonal profiles of seed loss caused by *L. occidentalis* have been determined previously (Bates *et al.* 2002b; Strong 2006; Strong *et al.* 1998), these studies concluded their research in late July or shortly thereafter. The 2010–2012 research was the first indication of substantial seed loss in August.

One feature of this late-season seed loss is that it is prevented by installing bags of polyester nylon mesh (1 mm mesh size) over the second-year cones. It has been known since 1994 that

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installing insect bags increases seedset in lodgepole pine (Strong *et al.* 1998). One reason mesh bags increase seedset is exclusion of the *L. occidentalis* (e.g., Bates *et al.* 2002a; Strong 2006; Strong *et al.* 2001). The possibility that mesh bags might also influence seedset by altering microclimate has never been rigorously addressed. Strong *et al.* (2001) tried to test microclimate effects by using different types of bags that either excluded or allowed entry of *L. occidentalis*, while varying the effects of temperature and relative humidity within the bag: it appeared that microclimate was not relevant to final seedset, while *L. occidentalis* exclusion was. Furthermore, excluding *L. occidentalis* by pesticides resulted in the same increase in seedset as protecting cones with mesh bags, suggesting that bag microclimate is unimportant (Strong *et al.* 2001).

Mesh-bag induced changes in the microclimate around the developing cone have never been directly measured. It has been theorized that seed orchards in the BC southern Interior are too hot and dry for good seed production (Dr. Michael Carlson³, personal communication), particularly considering that lodgepole pines typically grow in cooler, higher-elevation biogeoclimatic zones (Meidinger and Pojar 1991). Mesh bags used in these hot seed orchards may cool the cones or increase humidity, or both, thus preventing the observed seed loss. Damage attributed to excess temperature may be the result of a high mean temperature, time spent above a certain threshold temperature, or maximum temperature experienced in the seed-bearing portion of the cones. Increased humidity may reduce water stress inside cones.

Visual counts showed *L. occidentalis* densities to be low from 2010 to 2012 (Kalamalka Seed Orchards monitoring data, unpublished). This suggested that the prevention of August seed loss by mesh bags might be due to microclimate changes rather than *L. occidentalis* exclusion. The current experiment was designed to determine whether mesh bags increased seedset because of changes in temperature or humidity, or because of exclusion of *L. occidentalis*.

It has been found that *Leptoglossus occidentalis* prefers to feed on certain genotypes, termed 'clonal preference' (Blatt and Borden 1996, 1999; Richardson 2013). Certain genotypes were strongly favoured by *L. occidentalis*, while others were strongly avoided. Clonal preference was not stable from year to year, possibly because *L. occidentalis* were using host-finding cues that are not strongly genetically determined, and thus change yearly independent of clone. Host-selection cues might include cone size, cone count, or both (Blatt and Borden 1996, 1999; Richardson 2013); cone chemistry (Richardson 2013); or visual cues (Zahradnik 2012).

Some of these earlier studies were conducted in one of the two orchards used in the current study (Kalamalka Seed Orchards #307). Because clonal preference was not completely stable and the other orchard has no clonal-preference profile, I did not use preferred clones in this study. Similarly, we have no information on the influence of bag microclimate on clonal variation. Furthermore, I was interested in maximizing the generalizability of any conclusions. It was possible to use five ramets (individual trees) of one clone in one orchard, and five ramets of another clone in the other orchard (there is no overlap of clones between the two orchards). This would have reduced interplot variation for a more statistically powerful experiment at the expense of generalizability. Furthermore, any clone choice would have a good probability of selecting clones that were non-preferred, resulting in poor *L. occidentalis*-feeding data. So, to maximize generalizability and spread the risk of selecting non-preferred clones, 10 different clones were used in this study.

MATERIALS AND METHODS

In 2013, 10 lodgepole pine trees representing 10 different clones were randomly selected, five of each growing in each of two commercial seed orchards near Vernon, BC. Orchards were Orchard 307 (50° 14' 11" N, 119° 16' 39" W), with trees 30–32 years old and heights up to 8 m; and Orchard 339 (50° 23' 43" N, 119° 12' 31" W), with trees 10–12 years old and heights of 4–6 m. Trees were managed according to standard management of conifer seed orchards in BC,

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Table 1
Response variables measured on each of the four cone clusters selected on each experimental tree.

Cone cluster	Bagged	Response variable	Cones used
1	Yes	Temperature	2
		Humidity	--
2	No	Temperature	2
		Humidity	--
3	Yes	<i>Leptoglossus</i> feeding	3–4
4	No	<i>Leptoglossus</i> feeding	3–4

including drip irrigation, fertilizing, pruning, pest management, and harvest (Forest Genetics Council of BC 2014).

On each of the 10 trees, four cone clusters were selected, with three to four second-year cones in each. Cone clusters within each tree were of approximately equal aspect and height, and contained cones of approximately equal size. Cones and clusters were assigned to measuring response variables as described below (Table 1).

Internal cone temperatures: On each of the 10 selected trees, a 1-mm hole was drilled into two cones in each of two clusters (four cones per tree). Holes were drilled from near the stem end of the cone into the seed-bearing portion, about 1 cm deep. One K-type thermocouple with 30-gauge wires (0.26 mm diameter) was inserted into each hole (Fig. 1). Within one day, pitch had sealed off the hole and fixed the thermocouple wires in place. All four thermocouples on each tree were plugged into a battery-powered 4-channel thermocouple datalogger (model SD947, Reed Instruments, Ste. Anne de Bellevue, Quebec) that was programmed to record thermocouple temperatures every 5 minutes. One cone cluster with two thermocouple-containing cones was enclosed in a white polyester mesh bag (hole size: 0.6 x 0.9 mm); the other cluster was left unbagged. Thus, on each experimental tree, there were two cones inside mesh bags, and two outside, and the internal temperatures of each were measured every 5 minutes.

Mean internal cone temperatures, peak temperatures, and time above putative threshold temperatures of 35 and 40 °C were computed. Time above threshold temperatures was computed as degree-days by subtracting the threshold temperature from the measured temperature for each 5-minute interval, and summing positive differences. The temperature measurement for each 5-minute interval was assumed to be the mean temperature for that interval.

Data were analyzed in R (R Core Team 2013) by one-way RCB ANOVA (blocked by tree) after transformation with log(x+1) to normalize the residuals when applicable, for mean temperatures and cumulative degree-days, and by paired t-tests for peak temperatures and monthly degree-day accumulations.

Relative humidity (RH): To measure RH, weatherproof iButtons (Maxim Integrated, San Jose, CA) were used. These are about 17 mm diameter and 6 mm thick (Fig. 2), and can be preprogrammed to measure temperature and RH at desired intervals, then left in the field to run independently. iButtons were programmed to measure RH every 10 minutes, then one iButton was attached (using fabric first-aid tape) to a third cone in each of the cone clusters used for thermocouple measurements. The iButton was positioned on the lower north side of each cone to avoid sun as much as possible, and the RH-monitoring hole was positioned away from the cone and left exposed by the tape. Thus, on each experimental tree, RH around one bagged cone cluster was measured every 10 minutes, and RH around a non-bagged cone cluster was also measured the same way.

Mean daily RH was computed for the iButton inside and outside bags on each tree. Data were analyzed with a randomized complete block split-plot with trees as blocks, date as main plots, and



Figure 1. Thermocouple wire inserted into a 1-mm hole drilled into the seed-bearing portion of a lodgepole pine cone.

bagging (bagged or unbagged) as sub-plots. Missing data lead to an unbalanced design, so R package lmerTest (Kuznetsova *et al.* 2014) was used, which estimated the degrees of freedom for an unbalanced type III ANOVA.

Leptoglossus occidentalis feeding: The two remaining cone clusters on each tree (those not used for temperature and RH measurements) were used for measuring *L. occidentalis* feeding. On one cluster, a mesh bag was installed to exclude *L. occidentalis* from the cones. Pine needles and small branches surrounding the cone cluster held the bag away from the cones, preventing *L. occidentalis* from feeding through the bag. The second cone cluster was left unbagged. A time-lapse digital camera (Wingscapes Inc, Alabaster, AL; Fig. 3) was installed 1–1.5 m away and aimed at the unbagged cone cluster. Cameras were set to take one photo every 5 minutes from 06:00 through 22:30, starting May 13 and ending September 9. *Leptoglossus occidentalis* is known to be a diurnal insect (Koerber 1963; Hedlin *et al.* 1980), so photos were not taken at night.

All photos were examined for the presence of *L. occidentalis* adults and nymphs. Camera resolution was sufficient to distinguish between adults and nymphs, but not between male or female adults, nor between instars. Insect activity was classed as feeding, walking, or resting. *Leptoglossus occidentalis* feed by inserting their stylets into a developing seed within a cone, injecting salivary enzymes that macerate seed contents, and sucking the resulting liquid out (Koerber 1963). This process takes time, so if the insect—and particularly its head—remained in a similar position for >5 minutes, it was considered to be feeding. If the insect changed positions in consecutive photos, it was considered to be walking. Feeding *L. occidentalis* tend to move their bodies somewhat while holding their heads in approximately the same place: if an insect remained in exactly the same position, it was considered to be resting.

If a photo documented an *L. occidentalis*, the activity of the *L. occidentalis* was considered to have spanned a mean of 5 minutes. The time adults and nymphs spent feeding was summed for early season (May 13–June 30), late season (July 1–Sept 9), all season, and weekly.

After September 9, cones were collected from the bagged clusters and the exposed, photographed clusters. On collection, it was discovered that the exclusion bag for tree Kal 01 had been installed on the wrong tree, so tree Kal 01 was dropped from further analysis. Seeds were extracted at the Kalamalka Forestry Centre, Vernon, BC. Cones were dipped for about 10 s in water heated to between 90 and 96 °C to break any serotinous bonds between cone scales. Cones

were then baked for 8 h at 45 °C, then tumbled in a cone tumbling machine to dislodge seeds. Wings were removed by gently rubbing in a moistened cotton bag. Clean seeds were then X-rayed in a digital X-ray machine (Carestream InVivo DXS Pro, Woodbridge, Connecticut), and filled and empty seeds were counted. A seed was considered filled if the megagametophyte, embryo erosion cavity, and embryo were intact and filled at least four-fifths of the inside length of the seed coat (Kolotelo *et al.* 2001; Owens 2006). Most empty seeds were without megagametophytes, though remnants of the nucelli were apparent; occasionally, small, deformed, or eroded megagametophytes were present.

Seedset data were calculated as averages for each cone cluster: total seeds per cone, filled seeds per cone, and percent filled seeds. Seedset between bagged and unbagged cones was compared by paired t-test with bagged/unbagged for each tree, after transformation with $\log(x+1)$ to normalize the residuals when appropriate. Seed decline for each tree was computed as the difference in seedset data between the cones exposed to *L. occidentalis* feeding (unbagged cones) and cones on that tree protected from feeding (bagged cones). Seed decline was regressed against the cumulative number of minutes/cone/camera-day the exposed cones were fed upon by *L. occidentalis* for early season, late season, and all season.

RESULTS AND DISCUSSION

Internal cone temperatures: A total of 893,261 cone temperature measurements were taken between May 12 and September 5. Because of equipment malfunctions, about 443,000 potential measurements were missed. Equipment malfunctions were of a nature that missing data were never individual data points or small groups of data points, but whole days or multiple days. Because of this, it is not possible to extrapolate from adjacent data, and these data points were registered as missing data.

During early season (before July 15), mean cone temperatures of bagged cones (20.36 °C) were significantly higher than unbagged cones (20.01 °C; $F_{1, 196637} = 54.69$, $P < 0.0001$). During the late-season seed-loss period of July 15–Sept 6, mean bagged cone temperatures were also higher (23.38 °C) than unbagged (23.01 °C; $F_{1, 251911} = 56.62$, $P < 0.0001$). These figures represent



Figure 2. iButtons showing the RH-monitoring hole.



Figure 3. Time-lapse camera aimed at an unbagged cone cluster.

the average day and night temperatures. It is expected that the critical high temperatures would occur during daytime. It is possible that bags maintain cone warmth at night and provide cooling during the day, resulting in the observed slight mean warming. Therefore, means were computed for daytime (08:00 to 19:59) and night time (20:00 to 07:59) periods during late season, July 15–Sept 6. Daytime temperatures had an even greater differential in bagged vs. unbagged cones (28.92 and 28.05 °C, respectively; Fig 4A; $F_{1, 126089} = 453.9$, $P < 0.0001$). On the other hand, during the night, cone temperatures were slightly cooler inside the bags than outside (17.82 and 17.95 °C, respectively; Fig 4B; $F_{1, 125811} = 45.79$, $P < 0.0001$).

Peak internal cone temperatures were generally higher for bagged than unbagged cones (Table 2). Average peak temperature was significantly higher inside bags (43.14 °C) than outside (41.62 °C; $t = 2.44$, two-tailed $P = 0.0245$). Accumulated degree-days over 35 °C were significantly higher inside bags than outside (Fig 5A; $F_{1, 9} = 11.19$, $P = 0.00860$), and degree-days above 40 °C occurred almost exclusively in bagged cones (Fig. 5B; $F_{1, 9} = 4.82$, $P = 0.0557$).

It seems clear that the mesh bags did not cool cones as hypothesized, rather they increased cone temperature, especially during daytime. It is unlikely that heating the cones beyond the already hot Okanagan summers would somehow protect seeds, preventing the loss of seedset inside bags. Thus we infer that temperature is not the reason that mesh bags prevent seed loss.

Relative humidity: The iButtons were active for 10 days in late May and early June before they ceased collecting data. They were restarted on July 22, and they worked through August. In early season, there was no significant difference between RHs inside and outside bags, at 37.2% and 37.0%, respectively (Fig. 6; $F_{1, 86.9} = 0.2591$, $P = 0.612$). The later-season measurements, taken from July 22 to August 28, were from the seed-loss period of interest. During this time, the RH was significantly higher inside (53.0%) than outside (51.4%) bags (Fig. 6; $F_{1, 151} = 168.39$, $P < 0.0001$).

The late-season differences in RH were largely driven by high-RH (rainy) days. Days on which the bagged RH exceeded 60% ($n=13$) had a mean increase of 3.27% RH inside the bags, while days on which bagged RH was 59% or less had a mean increase of only 0.98% inside the bags. This could be because the bags retain moisture on foliage and cones longer after a rain event, leading to a longer period of elevated RH than unbagged cones. If moisture stress was a

limiting factor to seed development, then these humid days would be unimportant in seed loss; only the low-RH days would be important. But the difference in RH between bagged and unbagged cones during dry spells is less than 1%. It is difficult to envision this small difference being biologically relevant to seed health.

The biological relevance of increased humidity is particularly drawn into question when one considers that the variation in RH between trees is greater than the variation between bagged and unbagged clusters within a tree for any given date. Humidity inside bags is lower on some trees than humidity outside bags on other trees; this occurs for every single date after July 22. Even so, bagging increased seedset in every instance (see seed data below). If an increase of 1% RH were responsible for an increase in seedset, then we would have seen higher seedset in cone clusters with higher RH, regardless of bagging status. This was not the case. Therefore, we infer that relative humidity is not the reason that mesh bags prevent seed loss.

Leptoglossus occidentalis feeding: Over the course of the summer, 185,550 images were taken (e.g., Fig. 7). Feeding of adults and nymphs varied through the season (Fig. 8). Some cameras failed to function on some days, so feeding for each week is a mean of cameras operating during that week, expressed as minutes of feeding per camera-day. From Fig. 8, it is apparent that overwintered adults invaded the orchards through late May and June, and fed on developing cones. They oviposited for an extended period; their nymphs started to feed in late June. Feeding by overwintered adults declined as they died off by early July. Nymph feeding increased to a peak in late July. As nymphs matured, they emerged to the second-generation of adults, which started to feed in mid-July. Nymph and second-generation adult feeding declined through August, as nymphs matured to adults, and as adults left the orchard for overwinter sites. The bulk of feeding occurred during the time of interest, mid-July through August.

Cumulative *L. occidentalis* feeding was divided into early season and late season based on Fig. 8. The last feeding of overwintered adults occurred around June 30; this was also about the time

Table 2
Peak internal cone temperatures achieved (°C). Mean peak temperatures of bagged and unbagged cones were significantly different (T= 2.44, two-tailed P= 0.0245).

Tree	Bagged		Unbagged	
	T1	T2	T3	T4
ER 1	39.6	39.3	38.7	41.2
ER 2	39.4	41.8	38.7	42.2
ER 3	42.9	42.8	43.5	39.4
ER 4	46.9	43.7	39.2	46.8
ER 5	39.6	40.8	40.2	42
Kal 1	45.4	42.8	42.6	40
Kal 2	41	43.3	40.3	42
Kal 3	43.9	51.9	45.4	42.4
Kal 4	41.9	47	43.4	42.5
Kal 5	43.2	45.5	39.6	42.3
MEANS	43.14		41.62*	

nymphs began to feed. Early season was thus defined as May 15–June 30, late season as July 1–September 1. No feeding was detected after September 1.

In early season, mean daily feeding on each cone ranged from 0.00 to 11.53 minutes for adults, and 0.00 to 1.11 minutes for nymphs (Fig 9A). In late season, mean daily feeding on each cone ranged from 0.06 to 16.53 minutes for adults, and 1.49 to 52.97 minutes for nymphs (Fig 9B). Cumulatively, feeding on each cone ranged up to 7.33 hours in early season, and 27.28 hours in late season. *Leptoglossus occidentalis* are known seed feeders; their presence for so many hours on each cone cluster strongly indicates they are reducing the number of seeds inside those cones. Access to cones is prevented by the insect-exclusion bags, thus preventing *L. occidentalis* from reducing the seedset in cones enclosed by the bags.

Trees varied dramatically in the extent of feeding they suffered (Fig. 9). In early season, two trees (ER 05 and Kal 04) were completely avoided by *L. occidentalis*, while cones from only five trees were fed upon for more than 2 minutes/cone/day. Similar, though less dramatic, differences

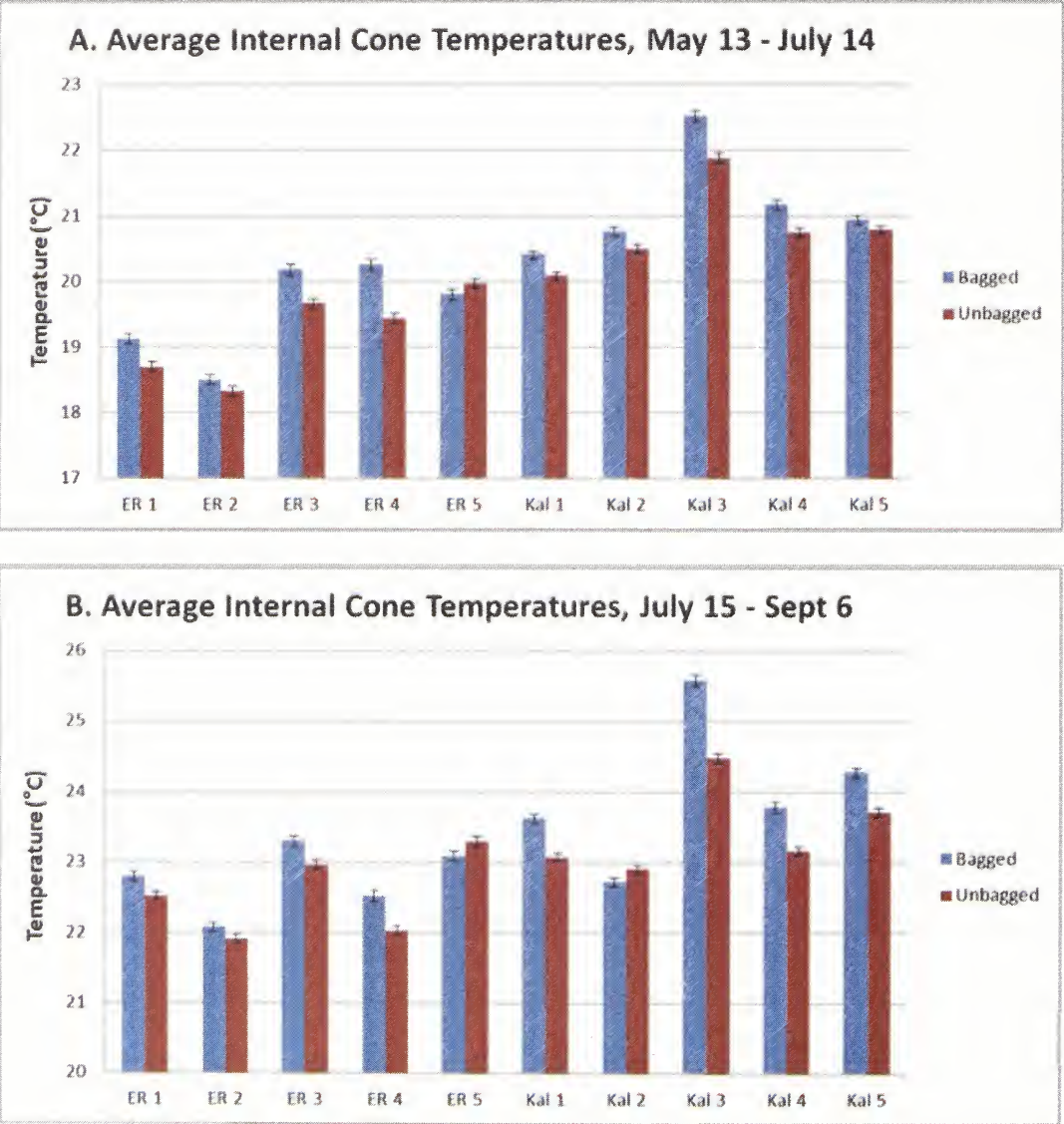


Figure 4. Mean internal cone temperatures (\pm SE) of bagged and unbagged cones in each of the 10 experimental trees. ER, Eagle Rock seed orchard 339. Kal, Kalamalka Seed Orchard 307.

exist for late-season feeding. These differences could be due to random variation or to preference for or avoidance of specific tree genotypes. The patterns found in this trial are consistent with the clonal preference findings of Blatt and Borden (1996, 1999) and Richardson (2013). Interestingly, the preferred or avoided genotypes changed from early season to late season, with the exception of ER 05, which was avoided in both time periods. It is possible that the host finding cues used by *L. occidentalis* change over time or that overwintered *L. occidentalis* use different foraging cues than second-generation *L. occidentalis*.

These foraging patterns also suggest that overwintered *L. occidentalis* prefer to feed on certain genotypes, while ovipositing on other genotypes. This is because nymphs are less mobile than adults: they are stuck on the trees their parents chose for oviposition. Nymphs were found on a different set of trees than 1st generation adults, so the adults must have oviposited on trees other than those they preferred for feeding. This in turn suggests there may be differences in foraging

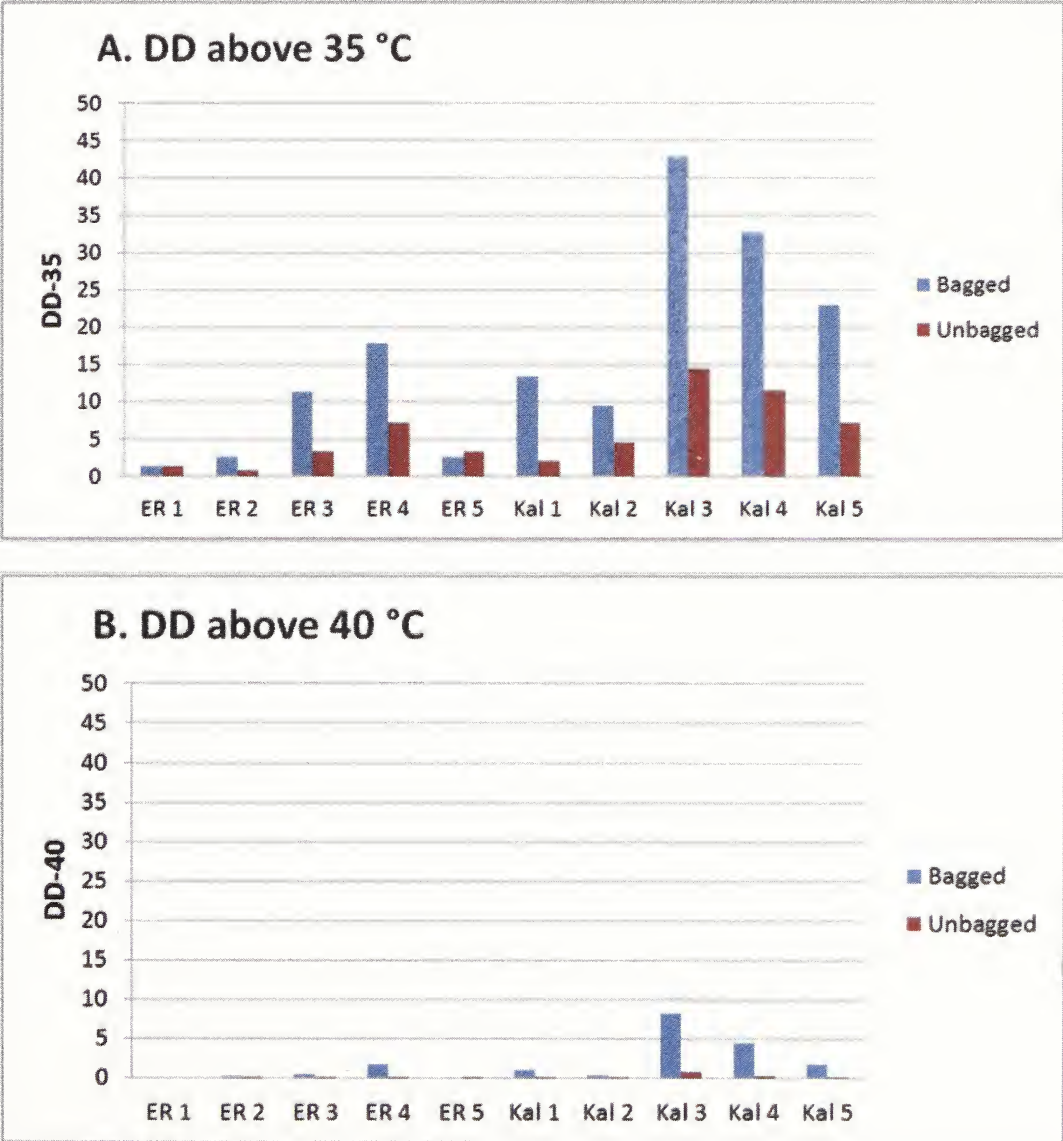


Figure 5. Degree-day sums of internal cone temperatures above a temperature threshold of 35 °C (A) or 40 °C (B).

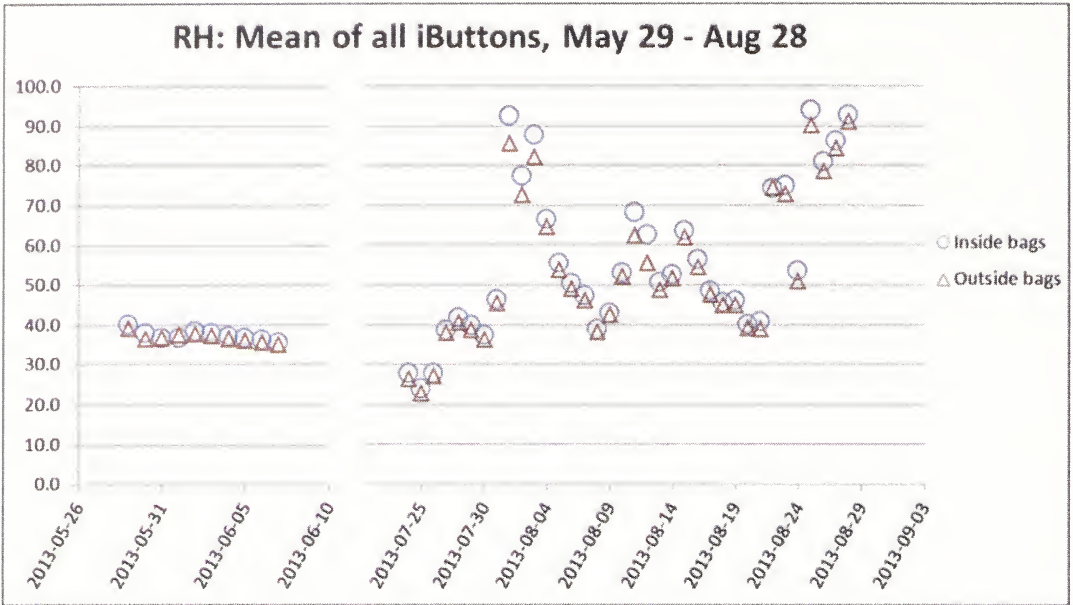


Figure 6. Daily relative humidity of cones inside and outside bags, averaged across all trees for each date.

cues used for feeding and oviposition. Different preferred hosts for adults and nymphs may be an adaptive behaviour, preventing depletion of nymph resources by adults.

Seed extractions. There was a mean of 11.8 filled seeds per cone inside the mesh bags, significantly ($P=0.0017$) more than the 2.8 filled seeds per cone found in unbagged cones (Fig. 10). Percent filled seed declined from 58.6% for bagged cones to 20.8% for unbagged ($P=0.0013$); total seeds per cone declined from 20.3 in bagged to 13.0 in unbagged cones ($P=0.018$). Preventing *L. occidentalis* access to cones within mesh bags thus appears to have prevented loss of seedset in the enclosed cones.



Figure 7. One image from a time-lapse camera, with a *L. occidentalis* visible on the centre cone.

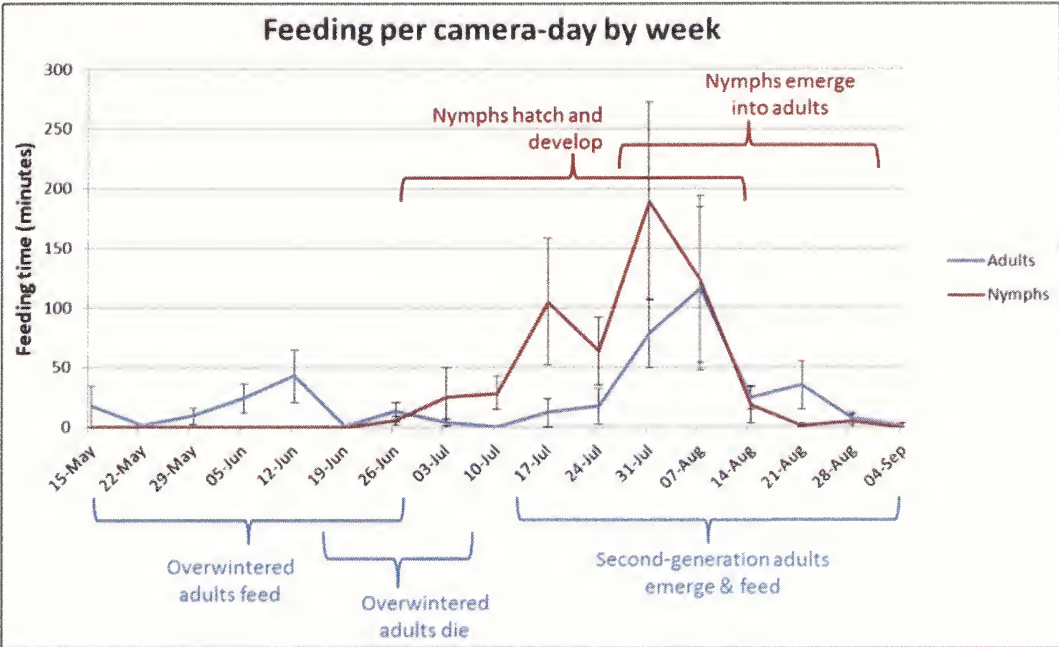


Figure 8. Minutes \pm SE of *L. occidentalis* feeding per camera-day for each week of the summer. Dates are mid-point of each calendar week. Insect phenology is demarcated for overwintered adults, nymphs, and second-generation adults. Standard errors reflect variation between trees in cumulative feeding during each week.

If feeding by *L. occidentalis* was responsible for the observed seed loss, then cones which experienced more hours of feeding should have sustained more seed loss. Seed loss was positively correlated to hours spent feeding on each cone cluster (Fig. 11). Regression analyses that were not forced through the origin all had intercepts not significantly different from zero, indicating that the main source of seed loss is *L. occidentalis* feeding. Because of this, and because theoretically there should be no seed loss without *L. occidentalis* feeding, all regressions were subsequently forced through the origin.

During July and August, the relationship between feeding and seed loss was strong ($r^2=0.6774$, $P=0.0035$; Fig 11A). An even closer relationship exists when feeding from May through August is considered ($r^2=0.8414$, $P<0.0001$; Fig 11B). These data suggest that *L. occidentalis* causes seed loss not only in late season, but in early season as well. This supports other research that has also found that *L. occidentalis* reduces filled seeds per cone by early-season feeding (Bates *et al.* 2002b; Strong 2006).

Percent filled seeds lost were strongly correlated with amount of feeding by *L. occidentalis* ($r^2=0.9306$, $P<0.0001$; Fig. 11C). Total seeds are included in the calculation for percent filled seeds, and total seeds are affected by early-season feeding (see below). Thus the season-long measurement for percent filled seeds is most relevant.

Total seeds lost was strongly related to amount of feeding in early season ($r^2=0.7389$, $P=0.0014$; Fig 11D). Early-season feeding has been shown to reduce total seeds per cone, since seeds fed upon prior to July 1 fuse to the cone scales and are not extracted (Bates *et al.* 2000). This trial confirms that early-season feeding reduces total seeds per cone.

CONCLUSION

This experiment conclusively proves that neither temperature nor relative humidity are the reason that enclosing second-year lodgepole pine cones in mesh bags increases seedset. Rather, the increased seedset is due in large part to protection from feeding by *L. occidentalis*. There may

be other minor factors associated with the mesh bags, such as direct insolation on the cones, reduced evaporative water loss from cones or nearby needles, or a combination of local factors that protect against whole-tree stress. However, the physiological mechanisms underlying any of these theories are unclear, and there is no evidence to even build a testable hypothesis based on these mechanisms. However, there is abundant and conclusive evidence that the increase in seed set created by enclosing cones in mesh bags is caused by *L. occidentalis* exclusion.

Leptoglossus occidentalis are not, however, the only cause of seed loss: they are the cause of the losses that are prevented by installing mesh bags. Even in the mesh bags, seed set at the site of this research is typically only 10–15 filled seeds per cone, with many empty seeds. This is much lower than the theoretical maximum of around 35 filled seeds per cone (Owens 2006), and lower than the seed set routinely found in cooler areas of BC, such as Prince George (which also has very few *L. occidentalis*). The poor seed set encountered even when *L. occidentalis* are excluded must be caused by other factors. Factors that have been or are being investigated include self-pollination, lack of pollen, pollination droplet problems, temperature and moisture relationships,

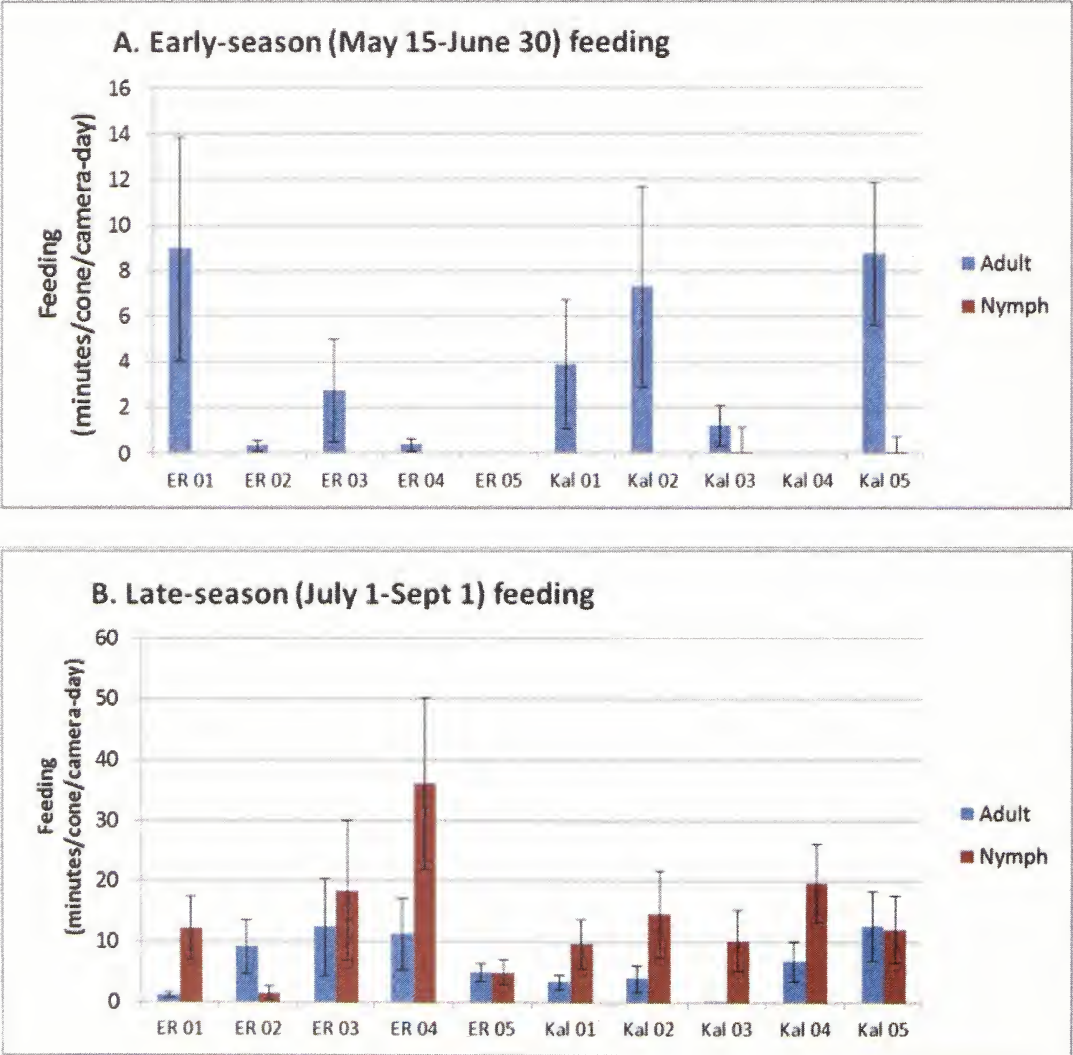


Figure 9. Mean feeding per cone per camera-day (\pm SE); early (A) and late (B) season for an exposed cluster on each of 10 trees. Standard errors reflect variation in daily feeding for each cone cluster; standard errors are large because no feeding occurred on most days for any given cone cluster.

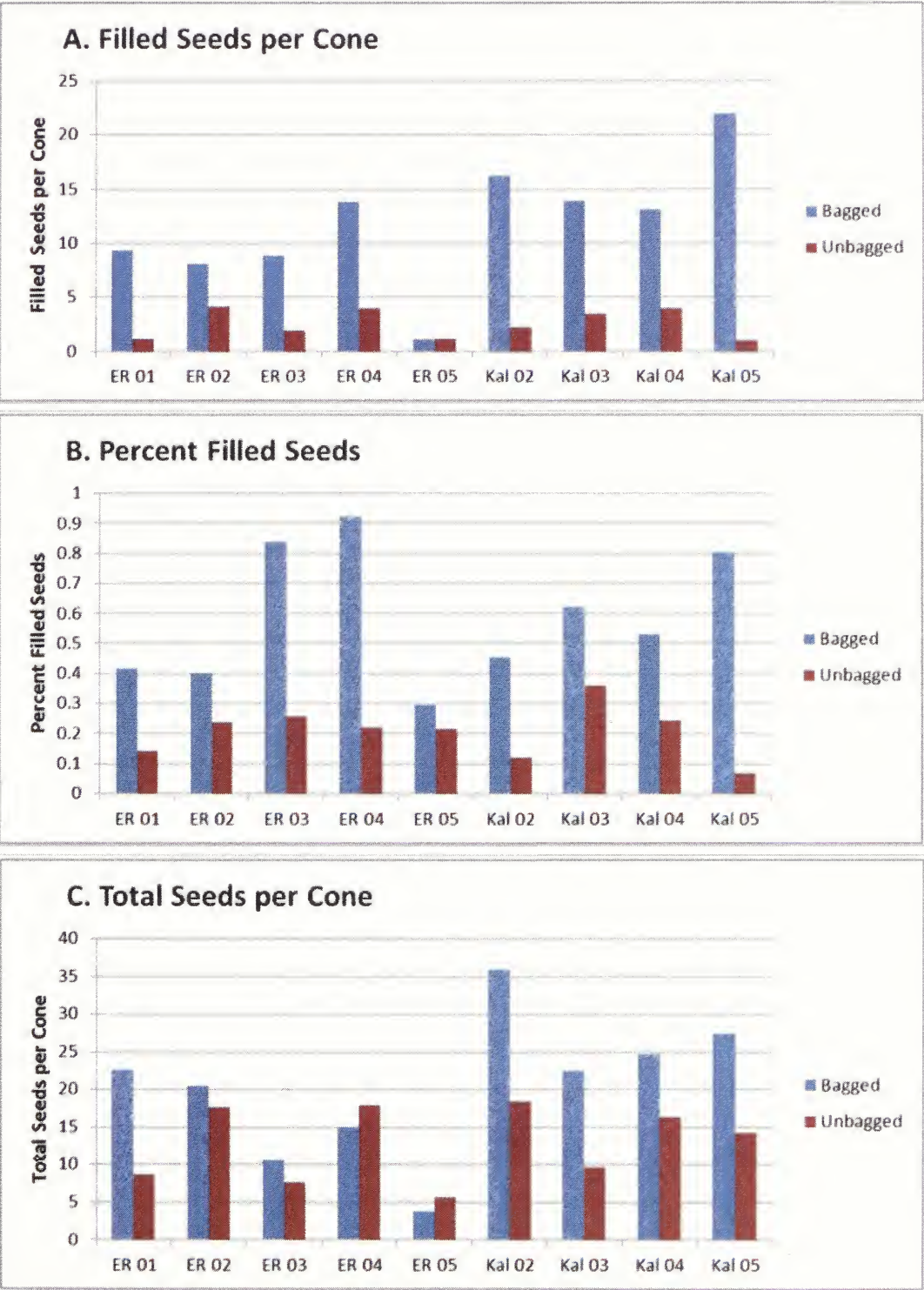


Figure 10. Seed extraction results for bagged and unbagged cones on each of 9 trees. A, mean Filled Seeds per Cone ($P= 0.0017$); B, mean Percent Filled Seeds ($P= 0.0013$); C, mean Total Seeds per Cone ($P= 0.0180$); all tests were 2-tailed paired T-tests.

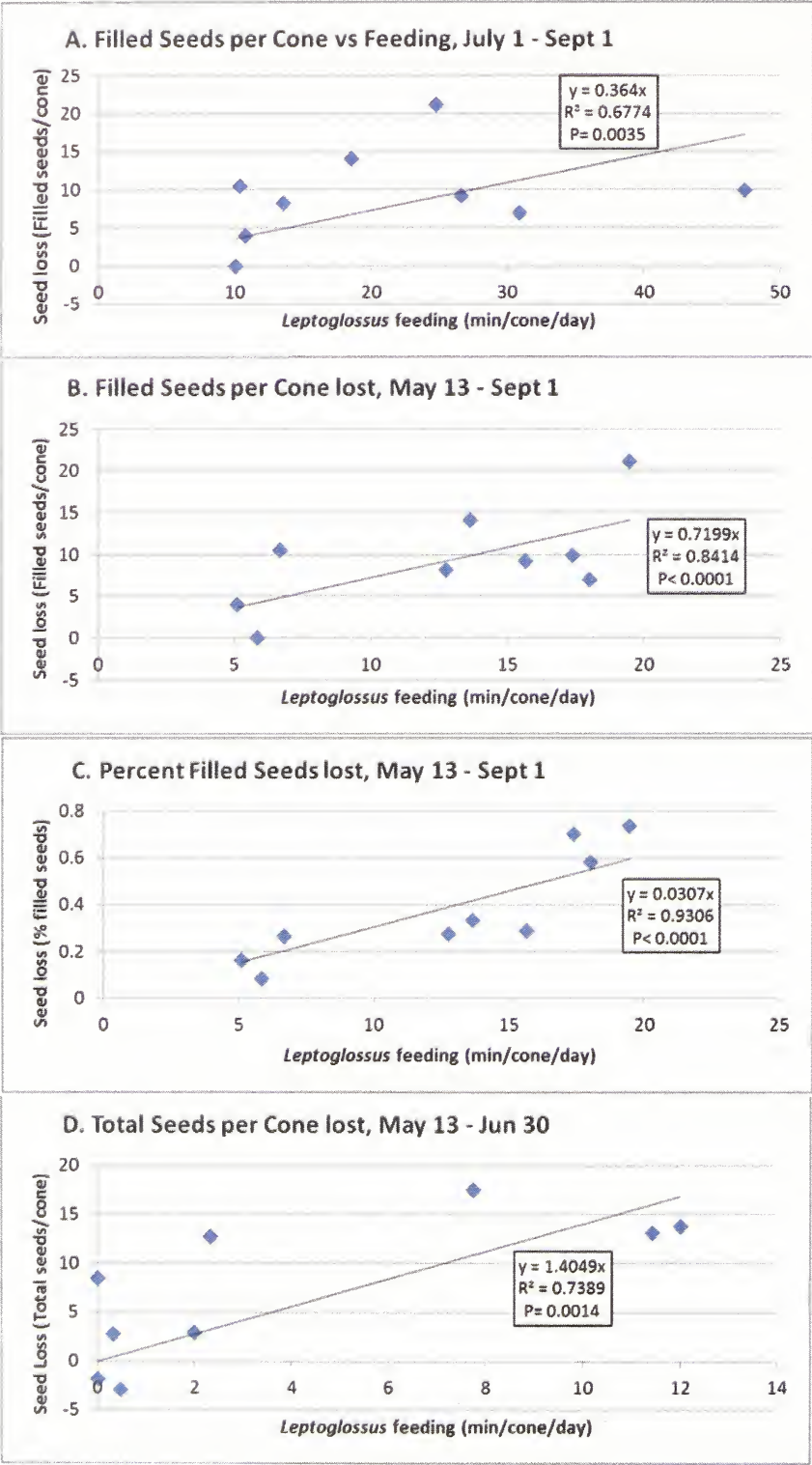


Figure 11. Seeds lost versus daily mean minutes of *L. occidentalis* feeding on each cone cluster. Feeding is combined feeding by adults and nymphs. Seed loss is calculated by subtracting seed values in unbagged cones from seed values in bagged cones.

other orchard culture factors, and seed fungal infections (Webber 2014; von Aderkas 2014). Despite years of research, however, the non-*L. occidentalis* causes of poor seed yield remain unknown.

Finally, despite using chemical control measures against *L. occidentalis*, lodgepole pine seed orchards in BC do still suffer from *L. occidentalis*-related seed loss (WBS, unpublished bagging trials in commercial seed orchards). Continuing research must identify how to manage *L. occidentalis* in order to prevent seed loss. Despite many years of research, we still have no efficient means of monitoring for *L. occidentalis*, no traps, no novel control tactics, and no registered pesticide. The insecticide Sevin XLR® (carbaryl) is registered against mountain pine beetles and pine sawflies, with poorly studied effects on *L. occidentalis*. A new pesticide (Matador®, lambda-cyhalothrin) is currently undergoing registration for *L. occidentalis* control, under the User-Requested Minor-Use Label program of the Pest Management Regulatory Agency (Caroline Bedard⁴, personal communication). This and a related pesticide are the main products used to control *L. occidentalis* in the USA; therefore, it may help manage seed loss due to *L. occidentalis* in Canada.

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Checklist of the Spheciform Wasps (Hymenoptera: Crabronidae & Sphecidae) of British Columbia

C. G. RATZLAFF¹

ABSTRACT

An annotated checklist of wasps in the families Crabronidae and Sphecidae is compiled for British Columbia, resulting in a total of 280 species, 70 of which are new to the province. In addition, 42 of the species reported here are new records for Canada, two species may be potentially new to science, and the European species *Ectemnius cephalotes* (Olivier) is recorded for the first time from western Canada. This study was done primarily through examination of museum holdings and a thorough review of existing literature. All available resources necessary for the identification of British Columbian taxa are cited. Intraprovincial distributions by ecoprovinces are presented for each species.

Key Words: Crabronidae, Sphecidae, Checklist, British Columbia

INTRODUCTION

The four currently recognized (i.e., Pulawski 2015) hymenopteran families of sphecoid wasps, Heterogynaidae, Ampulicidae, Sphecidae and Crabronidae, have, at different points in the past century, been treated as one family (e.g., Bohart and Menke 1976), as well as several families (Finnamore in Goulet and Huber 1993). Collectively, these families form the group known as the “spheciform”, “sphecoid”, or “apoid” wasps (Apoidea, Spheciformes); the historic Sphecoidea. Three of these families are found in North America, including parts of Canada, and two, Crabronidae and Sphecidae, occur in British Columbia.

The spheciform wasps have, historically, been poorly studied in British Columbia. In 1948, Spencer and Wellington published “A preliminary list of the Sphecinae of British Columbia”, the only provincial list of spheciform wasp species ever assembled. However, other provincial studies of insect fauna have included localized lists of spheciform wasps, but have almost exclusively been limited to the south Okanagan region of the province (Blades and Maier 1992; Scudder 1994). Buck’s 2004 checklist of Ontario spheciform wasps contains presence–absence data for some species in British Columbia, including multiple new records for the province. That said, there have been several notable contributions to the study of these wasps that have relevance to British Columbia. Valid name-bearing type specimens have been designated from British Columbia for ten species, consisting of six holotypes, two sets of syntypes, and two lectotypes. Paratypes for three valid species have also been designated from British Columbia. In addition, three holotypes and one set of syntypes for species that have since been synonymized are designated from British Columbia. The holotype for the species *Gorytes provancheri* (Handlirsch) was originally described in 1888 as *Gorytes laticinctus* by Provancher, which was a junior homonym of the European species *G. laticinctus* (Lepeletier 1832). Handlirsch redescribed the species in 1895, translating Provancher’s original description and naming it after the entomologist, before the type specimen was reported lost by Gahan and Rohwer (1917).

The size of spheciform wasps varies, ranging in British Columbia from 2 mm (*Ammoplanus* & *Parammoplanus*) to 30 mm (*Ammophila* & *Sphex*). Tremendous morphological variation in spheciform wasps exists at all taxonomic levels, and general characteristics are limited. A few useful family-level characteristics are the presence of lobes protruding posteriorly from the pronotum, having a pronotal collar with the posterior edge almost straight, and unbranched

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thoracic hairs. Members of the Family Sphecidae can usually be separated from other spheciforms by the presence of a petiole at the base of the abdomen consisting of only a sternal segment.

This checklist aims to provide a resource for future studies on spheciform wasps in British Columbia. It is hoped a provincial inventory will allow for easier identification of specimens and provide background information for environmental studies.

MATERIALS AND METHODS

The taxonomic classification followed for this checklist is Pulawski's (2015) Catalog of Sphecidae *sensu lato*.

Specimens examined and identified were from the following collections: Spencer Entomological Museum at the University of British Columbia (SEM), in Vancouver, BC, the Royal British Columbia Museum (RBCM), in Victoria, BC, the University of Northern British Columbia (UNBC), in Prince George, BC, the Canadian National Collection of Insects, Arachnids, and Nematodes (CNC), in Ottawa, ON, and the author's personal collection (CGR). In addition, electronic specimen records of spheciform wasps from British Columbia were received from Andrew Bennett at the CNC and Matthias Buck at the Royal Alberta Museum (PMAE). Krombein (1979) provided provincial records that served as a starting point for assembling the preliminary species list. Identification to the family level was facilitated using the book "Hymenoptera of the World" by Goulet and Huber (1993). Many of the families listed in A.T. Finnamore's "Series Spheciformes" section (1993) are currently subfamilies of Crabronidae, further aiding in identification. Identification to genus level followed Bohart and Menke (1976). Species identifications were done using primary literature whenever possible, including original species descriptions when necessary.

Only species with explicit records from British Columbia were included in this checklist (based on literature records or examination of actual specimens). A species was considered new to the province when no published record of that species from a locality in British Columbia was present in the literature. Species with published ranges such as "Transcontinental in Transition and Upper Austral Zones" (Krombein 1979) were included only when explicit specimen records were seen. Two doubtful species records were excluded from the list and are addressed after the checklist. Standard Canada Post and USPS abbreviations of provinces and territories in Canada and states in the United States are used in describing previously recorded distributions of newly recorded species.

Locality information from physical specimens, data provided by the aforementioned collections for material that was not examined, and literature records were used to assess the geographic distribution of the species within the province. Distributions were classified into ecoprovinces according to the ten terrestrial ecoprovinces represented in British Columbia (Demarchi 2011). The number of species recorded from each ecoprovince was totalled, and species distributions according to ecoprovince were compiled.

RESULTS AND DISCUSSION

As recognized here, there are nine subfamilies of spheciform wasps in British Columbia, represented by 64 genera and 280 species. The majority of these are Crabronidae, with 241 species in 55 genera and five subfamilies. Sphecidae is represented by four subfamilies, with 39 species in nine genera. There are 70 species (25.0%) newly recorded for British Columbia, 42 of which (15.0%) are also new to Canada. Two species potentially new to science were also identified, one in the genus *Miscophus* and the other in the genus *Nysson*; these will require further examination.

The total number of wasps from both families is summarized by ecoprovince in Figure 1. Lower numbers of species in the northern ecoprovinces, specifically the Southern Boreal Interior, are likely due to cooler climates, unsuitable for many species, and sampling bias towards more populated areas in the south.

The following descriptions are general summaries for each of the subfamilies and include nesting habits and provisioning information.

The Subfamilies of Crabronidae

Astatinae

Three genera and 16 species of astatine wasps are found in British Columbia. All species of *Astata*, *Diploplectron*, and *Dryudella* are groundnesting and provision their nests with heteropterans (Bohart and Menke 1976). Males of *Astata* and *Dryudella* possess holoptic eyes and are often seen perching on sticks or rocks. They have been observed making rapid flights around the immediate area and returning to the same spot facing the opposite direction (Parker 1962, 1969).

Bembicinae

Nineteen genera and 47 species of bembicine wasps are found in British Columbia. All species are groundnesting and most prefer habitats with sand or sandy soil, hence the common name of “sand wasps”. Four genera, *Bembix*, *Microbembex*, *Steniolia* and *Stictiella*, have been recorded nesting in aggregations (Bohart and Horning, Jr. 1971; Bohart and Gillaspay 1985). Aggregations of nesting *Bembix americana* Fabricius are often seen in the summer on hot sandy paths throughout the southern half of British Columbia. Most species provision their nests with Hemiptera (Auchenorrhyncha), with the exception of *Bembix*, *Glenostictia* and *Stenolia* (dipterans), *Stictiella* (lepidopterans), *Microbembex* (dead or dying arthropods), and *Epinysson*, *Hyponysson*, *Nysson*, *Stizoides* and *Zanysson* (cleptoparasites of other spheciform wasps, usually focusing on a single genus) (Bohart and Horning, Jr. 1971; Bohart and Menke 1976; Bohart and Gillaspay 1985).



Figure 1. Summarized distributions of wasps in the families Crabronidae and Sphecidae, by ecoprovince in British Columbia.
C = species in Crabronidae, S = species in Sphecidae

Crabroninae

The crabronine wasps are the most speciose and diverse subfamily in the province, with 17 genera and 95 species recorded. Most species are groundnesting (members of this subfamily are what are usually referred to as “digger wasps”) but some, e.g., members of the genera *Rhopalum* and *Solierella*, utilize existing cavities, such as holes in wood, stems or abandoned nests of other organisms (Bohart and Menke 1976). When provisioning their nest, each genus tends to have a preferred prey, but the variation between the genera is great. Twelve insect orders, primarily Diptera, Orthoptera and Hemiptera, as well as the arachnid order Araneae, have been recorded from the nests of Crabronine wasps (Williams 1960; Bohart 1976; Bohart and Menke 1976; Pulawski 1988). Members of the genus *Miscophus* are unusual in that they walk on the ground, resembling ants, while searching for prey; this mimicry provides them with protection from predators (Bohart and Menke 1976).

Pemphredoninae

Eleven genera and 55 species of this often-cryptic subfamily are recorded from British Columbia. Only nests of three genera, *Diodontus*, *Mimesa* and *Pulverro*, have been located exclusively in the ground (Bohart and Menke 1976; Finnamore 1983; Eighme 1989). All other genera have been found to nest primarily in holes in wood or in stems of plants. Thysanoptera and small Hemiptera (Auchenorrhyncha) are used exclusively to provision their nests. Four genera, *Diodontus*, *Passaloecus*, *Pemphredon* and *Stigmus*, prey exclusively on Aphididae (Bohart and Menke 1976; Eighme 1989). Many species in the subfamily are not commonly encountered, because of their small size. The genera *Ammoplanus* and *Parammoplanus* contain the smallest spheciform wasps in the province (~2mm). Their members, as well as those in *Pulverro*, *Spilomena* and *Stigmus*, are seldom over 5 mm in length and therefore rarely seen (Smith 2008).

Philanthinae

Five genera and 28 species of philanthine wasps are recorded from British Columbia. All species nest in the ground, and nest provisions for each genus are exclusively selected from a specific group of insects. *Aphilanthops* use winged ant queens of the genus *Formica*, *Cerceris* use adult Coleoptera primarily from the families Curculionidae and Buprestidae, *Clypeadon* exclusively use workers of the ant genus *Pogonomyrmex*, *Eucerceris* use adult Curculionidae, and *Philanthus* use a wide variety of bees as well as some wasps (Bohart and Grissell 1975).

The Subfamilies of Sphecidae

Ammophilinae

Two genera and 27 species are recorded from British Columbia. Species in the genus *Ammophila* provision their nest with the larvae of Lepidoptera and Hymenoptera (Symphyta), while species in the genus *Podalonia* primarily use larvae of the moth family Noctuidae. Females of *Podalonia* search on the ground for subterranean larvae, excavating those they find (Bohart and Menke 1976).

Chloriontinae

A single species, *Chlorion aerarium* Patton, has been newly recorded from the Okanagan–Similkameen region of British Columbia. This is a large metallic blue–green wasp that nests in the ground. They provision their nests with orthopterans primarily from the family Gryllidae, earning them the common name of “Steel-Blue Cricket Hunter” (Bohart and Menke 1976).

Sceliphrinae

Two genera, each with a single species, are recorded from British Columbia. The most common species, *Sceliphron caementarium* (Drury)—often known as a “mud-dauber”—is a medium-sized black and yellow wasp that constructs free nests out of mud. The second species, *Chalybion californicum* (de Saussure), is a similar-sized, metallic-blue wasp that nests in existing cavities, such as holes in wood or abandoned nests of other mud-daubers. Both species provision their nests with Araneae (Bohart and Menke 1976).

Sphecinae

Four genera and 9 species are recorded from British Columbia. Species in the genera *Palmodes*, *Prionyx*, and *Sphex* are groundnesting, while *Isodontia elegans* (Smith), the only representative of that genus in the province, nests in existing cavities. All species provision their nests with orthopterans, with *Isodontia* and *Sphex* almost exclusively using Gryllidae and Tettigoniidae, *Palmodes* exclusively using Tettigoniidae and Prophalangopsidae, and *Prionyx* exclusively using Acrididae (Bohart and Menke 1976).

Species Checklist

The following annotated checklist contains all of the species of Crabronidae and Sphecidae currently recorded from British Columbia. The “BC Reference” column contains any previously published reference to a British Columbian locality for that species. Species newly recorded by the author are noted. Species newly recorded by Matthias Buck, who has kindly allowed the author to present those findings here, are denoted by the symbol “*”. The symbol “†” denotes non-native species introduced to the province. Intraprovincial distributions according to ecoprovinces are listed for each species. The ecoprovinces included are the Georgia Depression (GD), Southern Interior (SI), Southern Interior Mountains (SIM), Coast and Mountains (CM), Central Interior (CI), Southern Boreal Interior (SBI), Northern Boreal Mountains (NBM), Boreal Plains (BP), and Taiga Plains (TP). No spheciform wasps have been recorded from the Southern Alaska Mountains ecoprovince, and therefore it was not included in the table. Species with distributions shown as “-” were recorded only as present in British Columbia by Krombein (1979) and only have “BC” listed as a location. Species-specific distribution information for newly recorded species is found in the New Records section following the checklist. Any other symbols refer to notes located at the end of the checklist. A version of this checklist is present on the E-Fauna BC website (www.efauna.bc.ca).

Table 1

An annotated checklist of all spheciform wasps recorded from British Columbia with intraprovincial distributions for each species.

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
Crabronidae: Astaninae										
<i>Astata bakeri</i> Parker	Parker (1962); Krombein (1979)	X	X							
<i>Astata bicolor</i> Say	New to BC			X						
<i>Astata leuthstromi</i> Ashmead	Parker (1962)		X	X			X	X		
<i>Astata mexicana</i> Cresson	Parker (1962); Krombein (1979)		X							
<i>Astata nevadica</i> Cresson	Parker (1962); Krombein (1979)		X							
<i>Astata nubecula</i> Cresson	Parker (1962)	X	X	X		X		X		
<i>Astata occidentalis</i> Cresson	Parker (1962); Blades and Maier (1992)		X	X						
<i>Astata unicolor</i> Say	Parker (1962)		X	X						
<i>Diploplectron brunneipes</i> (Cresson)	New to BC								X	
<i>Diploplectron fossor</i> Rohwer	Parker (1972); Krombein (1979)		X			X		X		
<i>Diploplectron peglowi</i> Krombein	Parker (1972); Blades and Maier (1992)		X			X				
<i>Dryudella caerulea</i> (Cresson)	New to BC	X	X	X						
<i>Dryudella immigrans</i> (Williams)	New to Canada		X							
<i>Dryudella montana</i> (Cresson)	New to Canada		X					X	X	
<i>Dryudella picta</i> (Kohl)	New to BC		X	X						
<i>Dryudella rhimpa</i> Parker	Parker (1969)	X	X							
Crabronidae: Bembicinae										
<i>Alysson guignardi</i> Provancher	Buck (2004)			X	X				X	
<i>Alysson radiatus</i> Fox	New to Canada		X							
<i>Alysson triangulifer</i> Provancher	Bradley (1920); Krombein (1979)	X		X	X	X		X		X
<i>Bembix americana</i> Fabricius	Krombein (1979); Buck (2004)	X	X	X	X	X	X			
<i>Bembix amoena</i> Handlirsch	Krombein (1979)		X	X		X				
<i>Bicyrtes ventralis</i> (Say)	Krombein (1979); Buck (2004)		X	X						

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
<i>Clitemnestra bipunctata</i> (Say)	New to BC	X	X	X						
<i>Didineis nodosa</i> Fox	Scudder (1994)	X	X	X						
<i>Epinysson mellipes</i> (Crcsson) ¹	Krombein (1979); Buck (2004)	-	-	-	-	-	-	-	-	-
<i>Epinysson moestus</i> (Cresson)	New to Canada*		X	X						
<i>Epinysson pacificus</i> (Rohwer)	New to Canada*		X							
<i>Glenostictia pulla</i> (Handlirsch)	New to Canada		X							
<i>Gorytes angustus</i> (Provancher)	Krombein (1979)		X	X						
<i>Gorytes atricornis</i> Packard	Krombein (1979); Buck (2004)	X	X	X						
<i>Gorytes canaliculatus</i> Packard	Krombein (1979); Buck (2004)			X		X				
<i>Gorytes flagellatus</i> Bohart	Krombein (1979)		X	X						
<i>Gorytes provancheri</i> Handlirsch	Provancher (1888); Krombein (1979)		X							
<i>Gorytes simillimus</i> Smith	Buck (2004)	X	X	X						
<i>Harpactus mendicus</i> (Handlirsch)	Handlirsch (1893); Krombein (1979)		X			X				
<i>Harpactus pictifrons</i> Fox	New to Canada		X					X		
<i>Harpactus rugulosus</i> (Bohart)	New to Canada		X							
<i>Hoplisoides hamatus</i> (Handlirsch)	Buck (2007)		X	X						
<i>Hoplisoides nebulosus spilopterus</i> (Handlirsch)	Buck (2007)		X	X	X					
<i>Hoplisoides punctifrons</i> (Cameron)	Buck (2007)		X							
<i>Hoplisoides tricolor</i> (Crcsson)	Buck (2007)		X							
<i>Hyponysson bicolor</i> Rohwer	New to Canada	X	X							
<i>Lestiphorus piceus</i> (Handlirsch)	Handlirsch (1888); Krombein (1979)	X		X						
<i>Microbembex californica</i> Bohart	Scudder (1994)	X	X							
<i>Microbembex nigrifrons</i> (Provancher)	New to Canada		X							
<i>Nysson chumash</i> Patc	New to Canada*	X		X						
<i>Nysson fidelis</i> Cresson	New to Canada*		X	X						
<i>Nysson gagates</i> Bradley	Bradley (1920); Krombein (1979)		X			X				
<i>Nysson hesperus</i> Bohart	Bohart (1968a); Krombein (1979)	X	X	X						X
<i>Nysson neorusticus</i> Bohart	New to Canada*	X		X						
<i>Nysson plagiatu</i> s Crcsson	New to BC		X							
<i>Nysson recticornis</i> Bradley	New to Canada*		X							
<i>Nysson tristis</i> Cresson	Krombein (1979)	X	X							
<i>Nysson sp.</i>	New to Canada*			X						
<i>Oryttus laminiferus</i> (Fox)	New to Canada		X							
<i>Oryttus mirandus</i> (Fox)	New to Canada		X							
<i>Steniolia obliqua</i> (Cresson)	Krombein (1979); Bohart and Gillasp (1985)		X							
<i>Steniolia scolopacea albicantia</i> Parker	Krombein (1979)		X			X				
<i>Steniolia tibialis</i> Handlirsch	New to Canada		X							
<i>Stictiella emarginata</i> (Crcsson)	Krombein (1979); Buck (2004)		X	X		X				
<i>Stictiella tuberculata</i> (Fox)	Bohart and Gillasp (1985); Scudder (1994)		X							
<i>Stizoides renicinctus</i> (Say)	Krombein (1979); Buck (2004)		X	X						
<i>Zanysson texanus fuscipes</i> (Crcsson)	New to Canada		X							

Crabronidae: Crabroninae

<i>Belomicrus columbianus</i> (Kohl)	Kohl (1892); Bohart (1994b)		X	X	X	X				
<i>Crabro advena</i> Smith	Bohart (1976); Lcclercq (2008)	X		X	X					
<i>Crabro argusinus</i> Bohart	Krombein (1979); Buck (2004)	X	X							
<i>Crabro conspicuus</i> Cresson	Bohart (1976); Lcclercq (2008)	X		X	X					
<i>Crabro flavicrus</i> Bohart	Bohart (1976)		X							
<i>Crabro florissantensis</i> Rohwer	Bohart (1976); Lcclercq (2008)		X	X	X					
<i>Crabro hispidus</i> Fox	Bohart (1976); Krombein (1979); Lcclercq (2008)		X							
<i>Crabro largior</i> Fox	Bohart (1976); Krombein (1979); Lcclercq (2008)		X	X	X					

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
<i>Crabro latipes</i> Smith	Bohart (1976); Krombein (1979); Leclercq (2008)	X	X	X	X			X		X
<i>Crabro monticola</i> (Packard)	Buck (2004)		X							
<i>Crabro nigrostriatus</i> Bohart	Bohart (1976)			X						
<i>Crabro pallidus</i> Fox	Bohart (1976); Leclercq (2008)							X		
<i>Crabro pleuralis</i> Fox	Fox (1895); Bohart (1976); Leclercq (2008)	X	X	X						
<i>Crabro tenuis</i> Fox	New to BC		X							
<i>Crabro vernalis</i> (Packard)	Bohart (1976); Leclercq (2008)							X		X
<i>Crossocerus angelicus</i> (Kincaid)	Krombein (1979); Leclercq (2000)	X		X	X	X				
<i>Crossocerus annulipes</i> (Lepeletier & Brullé)	Krombein (1979); Leclercq (2000)	X	X	X	X					
<i>Crossocerus barbipes</i> (Dahlbom)	Krombein (1979); Leclercq (2000)	X		X	X	X		X		
<i>Crossocerus chromatipus</i> Pate	New to Canada	X								
<i>Crossocerus elongatulus</i> (Vander Linden)	New to BC	X								
<i>Crossocerus harringtonii</i> (Fox)	Krombein (1979); Leclercq (2000)		X							
<i>Crossocerus lentus</i> (Fox)	Krombein (1979); Leclercq (2000)	X			X			X		
<i>Crossocerus leucostoma</i> (Linnaeus)	Bradley (1906); Leclercq (2000)	X	X	X	X	X		X		
<i>Crossocerus maculiclypeus</i> (Fox)	Krombein (1979); Leclercq (2000)	X	X	X	X	X		X		X
<i>Crossocerus maculipennis</i> (Smith)	Krombein (1979); Leclercq (2000)	X	X	X		X				X
<i>Crossocerus nigritus</i> (Lepeletier & Brullé)	Krombein (1979)	X	X	X	X	X				
<i>Crossocerus pseudochromatipus</i> Leclercq & Miller	Leclercq (2000)		X	X	X					
<i>Crossocerus stricklandi</i> Pate	Krombein (1979); Leclercq (2000)		X		X			X		
<i>Crossocerus tarsatus planipes</i> (Fox)	Krombein (1979); Leclercq (2000)	X		X	X	X		X	X	X
<i>Ectemnius alpheus</i> Pate	New to Canada		X							
<i>Ectemnius arcuatus</i> (Say)	Krombein (1979); Buck (2004)	X	X	X						
<i>Ectemnius atriceps</i> (Cresson)	Bohart and Kimsey (1979); Krombein (1979)	X	X	X						
<i>Ectemnius borealis</i> (Zetterstedt)	Buck (2004)	X	X	X	X	X		X		
<i>Ectemnius cephalotes</i> (Olivier) †	New to BC	X								
<i>Ectemnius continuus</i> (Fabricius)	Krombein (1979); Buck (2004)	X	X	X	X			X	X	
<i>Ectemnius dilectus</i> (Cresson)	Seudder (1994)		X	X		X				
<i>Ectemnius dives</i> (Lepeletier & Brullé)	Krombein (1979); Buck (2004)	X	X	X	X	X		X		
<i>Ectemnius lapidarius</i> (Panzer)	Krombein (1979); Buck (2004)	X	X	X	X	X		X		
<i>Ectemnius maculosus</i> (Gmelin)	New to BC	X	X							
<i>Ectemnius ruficornis</i> (Zetterstedt)	Krombein (1979); Buck (2004)	X	X	X	X	X	X	X	X	X
<i>Ectemnius rufifemur</i> (Packard)	New to BC		X	X						
<i>Ectemnius spiniferus</i> (Fox)	New to BC		X							
<i>Ectemnius trifasciatus</i> (Say)	Krombein (1979); Buck (2004)		X	X	X				X	
<i>Larropsis capax</i> (Fox)	Bohart and Bohart (1966); Krombein (1979)		X	X						
<i>Larropsis corrugata</i> (Bohart & Bohart)	Krombein (1979)		X							
<i>Larropsis distincta</i> (Smith)	Bohart and Bohart (1966); Krombein (1979)	X	X							
<i>Larropsis tenuicornis</i> (Smith)	New to Canada		X	X						
<i>Lestica confluenta</i> (Say)	Leclercq (2006)	X	X	X	X	X				
<i>Lestica producticollis</i> (Packard)	Leclercq (2006)	X	X	X					X	
<i>Lindenius columbianus</i> (Kohl)	Kohl (1892)		X	X						
<i>Liris argentatus</i> (Palisot de Beauvois)	Buck (2004)		X	X						
<i>Miscophus evansi</i> (Krombein)	Seudder (1994)		X							

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
<i>Miscophus</i> sp.	New to Canada		X							
<i>Oxybelus emarginatus</i> Say	Bohart and Schlinger (1957)		X	X		X				
<i>Oxybelus uniglutinis</i> (Linnaeus)	Bohart and Schlinger (1957)	X	X	X	X	X		X		
<i>Oxybelus ventralis</i> Fox	New to Canada		X							
<i>Pisonopsis birkmanni</i> Rohwer	New to Canada		X							
<i>Pisonopsis clypeata</i> Fox	New to Canada		X							
<i>Plenoculus davisii</i> Fox	Williams (1960); Krombein (1979)		X		X					
<i>Plenoculus propinquus</i> Fox	New to Canada			X						
<i>Rhopalum clavipes</i> (Linnaeus)	Blades and Maier (1992)	X	X	X	X	X				
<i>Rhopalum coarctatum</i> (Scopoli)	Leclercq (2002)	X			X					
<i>Rhopalum occidentale</i> (Fox)	Krombein (1979); Buck (2004)			X	X					
<i>Solierella affinis</i> (Rohwer)	New to Canada		X							
<i>Solierella peckhami</i> (Ashmead)	New to BC		X					X		
<i>Solierella plenoculoides similis</i> (Bridwell)	New to Canada	X	X	X	X					
<i>Solierella</i> sp. (incermis-group)	New to Canada*		X							
<i>Tachysphex aequalis</i> Fox	Pulawski (1988); Blades and Maier (1992)		X	X						
<i>Tachysphex aethiops</i> (Cresson)	Krombein (1979); Pulawski (1988)	X	X	X		X				
<i>Tachysphex alpestris</i> Rohwer	Krombein (1979); Pulawski (1988)	X	X	X						
<i>Tachysphex amplius</i> Fox	New to Canada		X							
<i>Tachysphex antennatus</i> Fox	Pulawski (1988); Buck (2004)		X							
<i>Tachysphex apicalis</i> Fox	Pulawski (1988); Scudder (1994)		X							
<i>Tachysphex ashmeadii</i> Fox	Pulawski (1988)		X							
<i>Tachysphex clarconis</i> Viereck	Krombein (1979); Pulawski (1988)	X	X							
<i>Tachysphex hopi</i> Pulawski	New to BC		X							
<i>Tachysphex montanus</i> (Cresson)	Fox,(1894a); Krombein (1979); Pulawski (1988)	X	X			X				
<i>Tachysphex mundus</i> Fox	New to BC		X							
<i>Tachysphex orestes</i> Pulawski	Pulawski (1988)		X							
<i>Tachysphex pauxillus</i> Fox	Krombein (1979); Pulawski (1988)		X	X						
<i>Tachysphex pompiliiformis</i> (Panzer)	Krombein (1979); Pulawski (1988)	X	X	X	X				X	
<i>Tachysphex psammobius</i> (Kohl)	Pulawski (1988)	X	X							
<i>Tachysphex semirufus</i> (Cresson)	Krombein (1979); Pulawski (1988); Buck (2004)		X	X		X				
<i>Tachysphex similis</i> Rohwer	Scudder (1994)		X							
<i>Tachysphex tarsatus</i> (Say)	Pulawski (1988); Blades and Maier (1992)		X	X				X		
<i>Tachysphex verticalis</i> Pulawski	New to Canada			X						
<i>Tachysphex williamsi</i> Bohart	New to BC		X							
<i>Tachytes distinctus</i> Smith	Krombein (1979); Bohart (1994a)		X							
<i>Tachytes nevadensis</i> Bohart	New to Canada		X							
<i>Tachytes pennsylvanicus</i> Banks	Krombein (1979)		X	X		X				
<i>Tachytes sayi</i> Banks	New to BC		X							
<i>Trypoxylon bidentatum</i> Fox	Buck (2004)	X		X						
<i>Trypoxylon frigidum</i> Smith	Sandhouse (1940); Krombein (1979)	X	X	X	X	X				
<i>Trypoxylon sculleni</i> Sandhouse	Sandhouse (1940); Krombein (1979)	X	X	X		X				
<i>Trypoxylon tridentatum</i> Packard	Sandhouse (1940); Krombein (1979)		X	X						

Crabroninae: Pemphredoninae

<i>Ammoplanus vanyumi</i> Pate	Smith (2008)			X						
<i>Diodontus adamsi</i> Titus	Buck (2004)	X	X	X				X		

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
<i>Diodontus argentinae</i> Rohwer	Eighme (1989)	X		X						
<i>Diodontus bidentatus</i> Rohwer	New to BC		X	X		X				
<i>Diodontus boharti</i> Eighme	Blades and Maier (1992)		X	X						
<i>Diodontus crassicornus</i> Viereck	New to Canada		X							
<i>Diodontus flavitarsis</i> Fox	New to BC		X	X		X				
<i>Diodontus fraternus</i> Rohwer	New to Canada		X							
<i>Diodontus leguminiferus</i> Cockerell	Scudder (1994)		X						X	
<i>Diodontus minutus</i> (Fabricius)	Eighme (1989); Buck (2004)		X							
<i>Diodontus occidentalis</i> Fox	New to Canada		X							
<i>Diodontus rugosus</i> Fox	Eighme (1989); Scudder (1994)		X							
<i>Diodontus striatus</i> (Mickel)	Eighme (1989); Scudder (1994)		X	X						
<i>Diodontus vallicolae</i> Rohwer	Eighme (1989)		X	X						
<i>Diodontus virginianus</i> Rohwer	New to BC		X							
<i>Mimesa cressonii</i> Packard	Krombein (1979); Finnamore (1982); Buck (2004)		X							
<i>Mimesa dawsoni</i> Mickel	New to BC*		X							
<i>Mimesa edentata</i> (Malloch)	Finnamore (1982)		X							
<i>Mimesa gregaria</i> (Fox)	Finnamore (1982); Buck (2004)	X	X	X	X	X		X		
<i>Mimesa lutaria</i> (Fabricius)	Finnamore (1982); Buck (2004)	X	X	X	X			X		X
<i>Mimesa pauper</i> Packard	Finnamore (1982); Buck (2004)	X	X	X	X	X		X		
<i>Mimesa sabina</i> Gittins	New to Canada		X							
<i>Mimesa senijextee</i> Finnamore	Finnamore (1982)	X	X	X						
<i>Mimesa simplex</i> (Malloch)	Finnamore (1982)	X	X	X		X				
<i>Mimesa uncinata</i> Cresson	Krombein (1979); Finnamore (1982)	X	X			X	X			
<i>Mimumesa atratina</i> (Morawitz)	Buck (2004)					X		X		X
<i>Mimumesa clypeata</i> (Fox)	New to BC	X	X	X		X				X
<i>Mimumesa mixta</i> (Fox)	Krombein (1979)	X	X	X	X					
<i>Mimumesa nigra</i> (Packard)	Krombein (1979); Buck (2004)		X		X					
<i>Parammoplanus apache</i> Pate	Blades and Maier (1992); Scudder (1994)		X	X						
<i>Parammoplanus olamentke</i> Pate	Blades and Maier (1992); Scudder (1994)		X	X						
<i>Passaloecus armeniaca</i> Cockerell	Krombein (1979)	-	-	-	-	-	-	-	-	-
<i>Passaloecus borealis</i> Dahlbom	Vincent (1978)		X	X						
<i>Passaloecus cuspidatus</i> Smith	Vincent (1978)	X	X	X	X	X	X	X	X	
<i>Passaloecus lineatus</i> Vincent	Vincent (1978)			X						
<i>Passaloecus melanocrus</i> Rohwer	Vincent (1978)			X						
<i>Passaloecus monilicornis</i> Dahlbom	Vincent (1978); Buck (2004)	X	X	X	X	X		X	X	
<i>Passaloecus patagiatus</i> Vincent	Vincent (1978)		X	X		X				
<i>Passaloecus singularis</i> Dahlbom †	Vincent (1978); Buck (2004)	X								
<i>Pemphredon baltica</i> Merisuo	Dollfuss (1995); Buck (2004)			X				X		
<i>Pemphredon confertum</i> Fox	Dollfuss (1995)		X	X						
<i>Pemphredon grinnelli</i> (Rohwer)	Krombein (1979); Blades and Maier (1992)	X	X	X						
<i>Pemphredon inornata</i> Say	Blades and Maier (1992); Dollfuss (1995)	X	X	X	X	X		X	X	X
<i>Pemphredon lethifer</i> (Shuckard)	Dollfuss (1995)	X	X							
<i>Pemphredon lugubris</i> (Fabricius)	Dollfuss (1995)	X	X	X	X	X		X		X
<i>Pemphredon montana</i> Dahlbom	Krombein (1979)	X	X	X	X	X		X		X
<i>Pemphredon nearctica</i> (Kohl)	Dollfuss (1995)	X	X	X	X	X				
<i>Pemphredon rileyi</i> Fox	Dollfuss (1995)	X								
<i>Psenulus frontalis</i> (Fox)	New to Canada		X	X						
<i>Pulverro columbianus</i> (Kohl)	Kohl (1890a); Krombein (1979)		X	X						
<i>Spilomena alboclypeata</i> Bradley	Bradley (1906); Krombein (1979)			X	X					
<i>Stigmus americanus</i> Packard	Krombein (1979)	X		X		X				
<i>Stigmus fraternus</i> Say	New to BC		X	X				X		
<i>Stigmus fulvipes</i> Fox	Krombein (1979)	X	X							
<i>Stigmus inordinatus</i> Fox	Krombein (1979)	X	X	X						

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
Crabronidae: Philanthinae										
<i>Aphilanthops frigidus</i> (Smith)	Bohart and Grissell (1975)		X	X						
<i>Aphilanthops subfrigidus</i> Dunning	Krombein (1979)	X	X	X		X				
<i>Cerceris aequalis</i> Provancher	Krombein (1979)		X	X		X				
<i>Cerceris californica</i> Cresson	Spencer (1957); Scullen (1965); Krombein (1979)		X							
<i>Cerceris calochorti</i> Rohwer	New to BC		X							
<i>Cerceris cockerelli</i> Viereck ²	Scullen (1965); Bohart and Grissell (1975)		X	X						
<i>Cerceris convergens</i> Viereck & Cockerell	Scullen (1965); Krombein (1979)		X							
<i>Cerceris crucis</i> Viereck & Cockerell	Krombein (1979); Buck (2004)		X							
<i>Cerceris echo</i> Mickel	Buck (2004)		X							
<i>Cerceris funipennis</i> Say ³	Scullen (1965); Buck (2004); Kimoto and Buck (2014)		X							
<i>Cerceris nigrescens</i> Smith	Spencer (1957); Scullen (1965); Krombein (1979)	X	X	X	X	X		X		
<i>Cerceris sexta</i> Say ⁴	Scullen (1965)		X							
<i>Cerceris sextoides</i> Banks	Spencer (1957); Krombein (1979)		X	X						
<i>Cerceris tepaneca</i> de Saussure	Scullen (1965)		X	X						
<i>Cerceris vanduzeei</i> Banks	Krombein (1979)		X	X						
<i>Clypeadon laticinctus</i> (Cresson)	Bohart and Grissell (1975)		X							
<i>Eucerceris flavocincta</i> Cresson	Spencer (1957); Krombein (1979)	X	X	X		X				
<i>Eucerceris vittatifrons</i> Cresson	Scudder (1994)		X							
<i>Philanthus bilunatus</i> Cresson	New to BC						X			
<i>Philanthus crabroniformis</i> Smith	Krombein (1979)	X	X	X	X	X				
<i>Philanthus gibbosus</i> (Fabricius)	Bohart and Grissell (1975); Krombein (1979)	X	X	X	X					
<i>Philanthus inversus</i> Patton	Bohart and Grissell (1975)		X	X						
<i>Philanthus multimaculatus</i> Cameron	Krombein (1979)		X	X						
<i>Philanthus pacificus</i> Cresson	Bohart and Grissell (1975)	X	X	X						
<i>Philanthus pulcher</i> Dalla Torre	Bohart and Grissell (1975)		X	X						
<i>Philanthus ventrilabris</i> Fabricius	Bohart and Grissell (1975); Krombein (1979)		X	X						
<i>Philanthus ventralis</i> (Mickcl)	Bohart and Grissell (1975)		X			X				
<i>Philanthus zebratus</i> Cresson	Banks (1913); Krombein (1979)		X	X		X				
Sphecidae: Ammophilinae										
<i>Ammophila aberti</i> Haldeman	Spencer and Wellington (1948); Blades and Maier (1992); Scudder (1994)		X							
<i>Ammophila acuta</i> (Fernald)	New to Canada		X	X						
<i>Ammophila azteca</i> Cameron	Krombein (1979)	X	X			X				X
<i>Ammophila cleopatras</i> Menck	Buck (2004)		X							
<i>Ammophila extremitata</i> Cresson	Blades and Maier (1992); Scudder (1994)		X			X				
<i>Ammophila ferruginosa</i> Cresson	New to BC		X							
<i>Ammophila karenae</i> Menck	New to Canada		X							
<i>Ammophila kennedyi</i> (Murray)	Krombein (1979)		X	X	X	X				
<i>Ammophila macra</i> Cresson	Menck (1965)		X	X	X					
<i>Ammophila mcclayi</i> Menck	New to Canada		X							
<i>Ammophila mediata</i> Cresson	Krombein (1979); Buck (2004)	X	X	X	X	X	X	X	X	
<i>Ammophila placida</i> Smith	Spencer and Wellington (1948)		X			X				
<i>Ammophila polita</i> Cresson	Spencer and Wellington (1948)					X				
<i>Ammophila procera</i> Dahlbom	Spencer and Wellington (1948); Buck (2004)		X	X	X	X				
<i>Ammophila strenua</i> Cresson	Krombein (1979)	X	X							
<i>Podalonia argentifrons</i> (Cresson)	Krombein (1979)	-	-	-	-	-	-	-	-	-
<i>Podalonia atriceps</i> (Smith) ⁵	Spencer and Wellington (1948)	X	X	X		X		X	X	

Species Name	BC Reference	GD	SI	SIM	CM	CI	SBI	NBM	BP	TP
<i>Podalonia clypeata</i> Murray	New to Canada		X							
<i>Podalonia luctuosa</i> (Smith) ⁵	Spencer and Wellington (1948); Buck (2004)	X	X	X		X		X	X	
<i>Podalonia melaena</i> Murray	Spencer and Wellington (1948)		X	X						
<i>Podalonia mexicana</i> (de Saussure)	Spencer and Wellington (1948)		X	X		X				
<i>Podalonia mickeli</i> Murray	Murray (1940); Krombein (1979)		X	X						
<i>Podalonia occidentalis</i> Murray	New to BC		X	X						
<i>Podalonia robusta</i> (Cresson)	Spencer and Wellington (1948); Buck (2004)	X	X	X	X	X		X	X	
<i>Podalonia sericea</i> Murray	Spencer and Wellington (1948); Krombein (1979)		X	X						
<i>Podalonia sonorensis</i> (Cameron)	Scudder (1994)		X							
<i>Podalonia valida</i> (Cresson)	Spencer and Wellington (1948)		X			X				
Sphecidae: Chloriontinae										
<i>Chlorion aerarium</i> Patton	New to BC		X							
Sphecidae: Sceliphrinae										
<i>Chalybion californicum</i> (de Saussure)	Spencer and Wellington (1948); Buck (2004)	X	X	X						
<i>Sceliphron caementarium</i> (Drury)	Spencer and Wellington (1948); Buck (2004)	X	X	X	X	X	X			
Sphecidae: Sphecinae										
<i>Isodontia elegans</i> (Smith)	Spencer and Wellington (1948)		X							
<i>Palmodes californicus</i> Bohart & Menke	Krombein (1979)			X						
<i>Palmodes carbo</i> Bohart & Menke	Krombein (1979)	X	X	X		X				
<i>Palmodes hesperus</i> Bohart & Menke	Krombein (1979)		X							
<i>Palmodes laeiventris</i> (Cresson)	Spencer and Wellington (1948)		X							
<i>Prionyx atratus</i> (Lepelletier)	Spencer and Wellington, (1948); Bohart and Menke (1963); Buck (2004)		X							
<i>Prionyx canadensis</i> (Provancher)	Kohl (1890b); Spencer and Wellington (1948); Buck (2004)	X	X	X						
<i>Sphex ichneumoneus</i> (Linnaeus)	Buck (2004)		X	X						
<i>Sphex lucae</i> de Saussure	Spencer and Wellington (1948)		X	X						

¹ –*E. mellipes* (Cresson) is listed as recorded from British Columbia in Krombein (1979), but due to the taxonomic uncertainty of species in the genus, it should be considered with reservations (as per M. Buck, personal communication).
² –*C. cockerelli* Viereck is listed and described incorrectly as *C. acanthophila* Cockerell in both Scullen (1965) and Bohart and Grissell (1975) (Ferguson 1983).
³ –*C. fumipennis* was recently rediscovered in Merritt and Lillooet, after not being recorded from the province since 1935 (Kimoto and Buck 2014).
⁴ –A specimen of *C. sexta* Say from British Columbia is listed as *C. stigmatialis* Banks in Scullen (1965). The specimen was originally determined as *C. fugatrix* Mickel by Scullen in 1957 (CNC specimen examined by the author), now considered a synonym of *C. sexta* Say (Ferguson 1984). Scullen (1965) incorrectly synonymized *C. fugatrix* Mickel with *C. stigmatialis* Banks, which is itself a synonym of *C. halone* Banks, an eastern Nearctic species that does not occur in British Columbia.
⁵ –The species recognition characteristics used by Murray (1940) to separate the two species *P. atriceps* (Smith) and *P. luctuosa* (Smith) are unreliable. Males can only be reliably identified using genitalia, and no diagnostic characteristic is known for the females (M. Buck, personal communication). Therefore, the intraprovincial distributions have been combined and used for both species.

New Species Records
Crabronidae: Astatinae
Astata bicolor Say

1 spm, Wycliffe, 25.vii.1928 (A. A. Dennys) [RBCM]

Distribution: Species new to BC. Previously recorded from Canada and the U.S. east of the Rockies (Parker 1962).

Diploplectron brunniepes (Cresson)

1♂, Peace R., 4 km E of Halfway R., 26.vii.1986 (S. G. Cannings) [SEM]

Distribution: Species new to BC. Previously recorded from AB, ID, CA, WY, and CO (Strickland 1947; Parker 1972).

Dryudella caerulea (Cresson)

1♂, Victoria, 18.viii.1906 [RBCM]; 1♂, Langford, 4.vii.1952, on sand (D. Evans) [CNC]; 1♂, Robson, 30.vii.1968 (H. R. Foxlee) [SEM]; 1♂, Robson, 8.viii.1968 (H. R. Foxlee) [SEM]; 1♂, Robson, 12.viii.1968 (H. R. Foxlee) [SEM]; 1♂, Kikomun Cr., 11.viii.1982 (R. A. Cannings & S. G. Cannings) [SEM]; 1♂, Comox, 16.viii.1987, sand pit (S. G. Cannings) [SEM]; 2 spms, Lytton, 0.5 km S on Hwy. 1, Lytton Cr., 50°13'23"N 121°34'28"W, 3.vii.1988, 1000' (G. E. Hutchings) [RBCM]; 1♀, Hornby Is., Norman Pt., 6.viii.1988 (S. G. Cannings) [SEM]; 1♂, Comox, 8.vii.1989, sand pit (S. G. Cannings) [SEM]; 1 spm, Summerland, Gartrell Point, 29.vi.1990, 49°34'22"N 119°37'06"W, 1140' (R. A. Cannings & H. Nadel) [RBCM]; 1♂ 1♀, Galiano Is., Bodega Ridge, 1.viii.1990, Manzanita/Douglas Fir, Malaise trap (S. G. Cannings) [SEM]; 1♀, Metchosin, Camas Hill, summit, 43°53'27"N 123°35'44"W, 8–15.viii.1999 (L. & C. Rosenblood) [RBCM]; 1♂, Saturna Is., Gulf Islands Nat. Pk., East Pt., 48.7846, –123.0449, 18.vii.2015 (C. G. Ratzlaff) [SEM]; 1♂, Saturna Is., Haggis Farm, Lyall L., 48.7871, –123.1600, 19.vii.2015 (N. Lee) [SEM]

Distribution: Species new to BC. Previously recorded from AB, WA, ID, CA, NV, CO, AZ, TX, and Mexico (Strickland 1947; Parker 1969; Krombein 1979).

Dryudella immigrans (Williams)

1♂, Lillooet, 1.viii.1917, sand (A. W. A. Phair) [RBCM]; 1♂, Penticton, West Bench, 5.vi.1983, rangeland/orchard edge, Malaise trap (S. G. Cannings) [SEM]; 1♂, Penticton, West Bench, 14.vi.1987, grassland/orchard edge, Malaise trap (S. G. Cannings) [SEM]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, NV, UT, WY, AZ, NM, TX, and Mexico (Parker 1969; Krombein 1979).

Dryudella montana (Cresson)

1 spm, Aspen Grove, 7.vii.1933 (K. Graham) [CNC]; 1 spm, Keremeos, 16.vii.1923 (C. B. Garrett) [CNC]; 1♀, Chilcotin, 12.vii.1930 (G. J. Spencer) [SEM]; 1♂, Nicola, 31.vii.1938 (G. J. Spencer) [SEM]; 2 spms, Taylor, 3.vii.1948 (Mason & Hughes) [CNC]; 1♂, Nicola, 30.vii.1944 (G. J. Spencer) [SEM]; 2 spms, Telegraph Cr., 3 mi. NE, 1.vii.1960 (R. J. Pilfrey) [CNC]; 1 spm, Ashcroft, Hat Creek, Finney Creek, 50°45'37"N 121°35'21"W, 15.vi.1989, sage flats near abandoned airstrip (R. A. Cannings & C. S. Guppy) [RBCM]; 1 spm, Ashcroft, Hat Creek, Finney Creek, 50°45'37"N 121°35'21"W, 13.vii.1989, sage flats near abandoned airstrip, Malaise (R. A. Cannings & C. S. Guppy) [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, UT, WY, and CO (Parker 1969; Krombein 1979).

Dryudella picta (Kohl)

1 spm, Kaslo, 30.vii.1906 (J. M. Cockle) [CNC]; 1 spm, Summerland, 9.viii.1916 (Sladen) [CNC]; 1♀, Kamloops, 26.vi.1938 (G. J. Spencer) [SEM]; 1♂, Robson, 21.vii.1957 (H. R. Foxlee) [SEM]; 1♂, Penticton, West Bench, 16.vii.1988, Malaise trap (R. A. Cannings) [RBCM]

Distribution: Species new to BC. Previously recorded from AB (Strickland 1947: as *Astata aspera* Fox), NT, ID, OR, CA, UT, CO, AZ, NC, and Mexico (Parker 1969; Krombein 1979; J. Ascher, Discoverlife).

Crabronidae: Bembicinae*Alysson radiatus* Fox

1♂, Oliver, 26.v.1945 (D. Blair) [SEM]; 1 spm, Osoyoos, Richter Pass, 24.v.1959 (L. A. Kelton) [CNC]; 1♂, Penticton, West bench, 5.vi.1983, rangeland/orchard edge, Malaise trap (S. G. Cannings) [SEM]

Distribution: Species new to Canada. Previously recorded from WA, CA, NV, and CO (Krombein 1979).

Clitemnestra bipunctata (Say)

15 spms, Kaslo, 3.viii.1916 (Sladen) [CNC]; 1 spm, Revelstoke, 28.vi.1952 (G. J. Spencer) [CNC]; 1 spm, Trail, 21.vii.1959 (L. A. Kelton) [CNC]; 1♀, Robson, 12.vii.1961 (H. R. Foxlee) [SEM]; 1♂, Robson, 16.viii.1963 (H. R. Foxlee) [SEM]; 1♂, Robson, 7.ix.1963 (H. R. Foxlee) [SEM]; 1♂ 3♀, Oliver, UBC Geology Camp, 20–23.vii.1989, pine/thicket edge, Malaise trap (S.G. Cannings) [SEM]; 1♀, Oliver, UBC Geology Camp, 24.vii.1989, at light (S. G. Cannings) [SEM]; 1♂, Summerland, Agriculture Canada Research Station, grassland research site, 25.vi.1990 (S. G. Cannings) [SEM]; 1♀, Summerland, Agriculture Canada Research Station, grassland research site, 27.vi.1990 (S. G. Cannings) [SEM]; 1♂, Oliver, UBC Geology Camp, 21.vii.1990, ponderosa pine forest, Malaise trap (S. G. Cannings) [SEM]; 1♂, Oliver, UBC Geology Camp, 22.vii.1990, hawthorn thicket, Malaise trap (S. G. Cannings) [SEM]; 1♀, Vancouver I., Saanich, Mt. Newton, 25.vi–2.vii.1993, FC orchard, Malaise (D. Blades) [RBCM]; 1♀, Oliver, Wolf Creek, Manual Flats, 49°11'47.76" N 119°30'46.89" W, 27.vii.1997, 429m (G. G. E. Scudder) [RBCM]; 1♂, Kinbasket Reservoir, Bush Arm (approx. 60 km NNW of Golden), 474424 5738727, 10.vii.2010, 753m (V. Prigmore & V. Smith) [RBCM]

Distribution: Genus new to BC. Previously recorded from AB, ON, QC (Buck 2004), and the Upper and Lower Austral Zones of the U.S. (Krombein 1979).

Epinysson moestus (Cresson)

1♀, Robson, 27.viii.1963 (H. R. Foxlee) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 27.vi.1981 (S. G. Cannings) [SEM]; 1♀, Okanagan, viii, (no collector) [CNC]

Distribution: Species new to Canada. Previously recorded from WA and CA (Krombein 1979).

Epinysson pacificus (Rohwer)

1♀, Osoyoos, Haynes Ecological Reserve, 10.vii–14.viii.1986, sand/*Artemisia/Purshia*, pitfall (S. G. Cannings) [SEM]; 1♀, Osoyoos, Haynes Ecological reserve, 14.vi–3.viii.1987, *Purshia/Aristida* steppe, pan trap (S. G. Cannings) [SEM]; 2♂, Osoyoos, Mt. Kobau, 8.vii–13.vii.1991 and 24.viii–28.viii.1991, 560m (D. Blades & C. Maier) [RBCM]; 1♀, Osoyoos, Haynes Ecological Reserve, 9.vii–7.viii.1994, BGxh1, pitfall trap (G. G. E. Scudder) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 13.vii–9.viii.1995, BGxh1, pitfall trap (G.G.E. Scudder) [SEM]; 1♂, Kilpoola L., 8.viii.1997 (G. G. E. Scudder) [SEM]

Distribution: Species new to Canada. Previously recorded from CA (Krombein 1979).

Glenostictia pulla (Handlirsch)

1♂ 1♀, Osoyoos, N end of Osoyoos L., 18.vii.1981 (R. A. Cannings) [RBCM]; 1♂, Osoyoos, Road #22, 18.vii.1981 (R. A. Cannings) [RBCM]; 1♂, Osoyoos, Haynes Ecological Reserve, lower sandy road, 20.vi.1989 (D. L. Sanford) [SEM]; 1♂, Osoyoos, Haynes Ecological Reserve, 49°04'49"N 119°30'46"W, 3.vii.1990 (R. A. Cannings & H. Nadel) [RBCM]

Distribution: Genus and species new to Canada. Previously recorded from WA, ID, OR, CA, and NV (Bohart and Gillaspay 1985).

Harpactus pictifrons Fox

3♂ 2♀, Penticton, West Bench, 4.iv.1983, rangeland/orchard edge, Malaise trap (R. A. Cannings) [RBCM]; 2♀, Telegraph Cr., 21.vi.1985, S-facing slope, pitfall trap (E. Krebs & J. J. Robinson) [SEM]; 1♂, Penticton, West Bench, 20.v.1987 (R. A. Cannings) [RBCM]; 4♂, Penticton, West Bench, 14.vi.1987, grassland/orchard edge, Malaise trap (S. G. Cannings) [SEM]; 1♂, Penticton, West Bench, 8.vi.1988, grassland/rose thicket edge, Malaise trap (S. G. Cannings) [SEM]; 1♂ 1♀, Penticton, West Bench, 24.vi.1988, grassland/orchard edge, Malaise trap (S. G. Cannings) [SEM]; 1♀, Hat Creek Valley, 4 km E of Finney L., Finney Creek, 15.vi–13.vii.1989, sage flats, Malaise (R. A. Cannings & C. Guppy) [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, ID, CA, UT, WY, CO, and AZ (Bohart 1980; Krombein 1979).

Harpactus rugulosus (Bohart)

1♂, Penticton, West Bench, 24.vi.1988, grassland/orchard edge, Malaise trap (S. G. Cannings) [SEM]; 2♀, Osoyoos, H.E.R., 13.vii–17.viii.1988, *Purshia/Aristida* shrub steppe, pitfall trap (S. G. Cannings) [SEM]; 1♀, Osoyoos, H.E.R., 9.vii–7.viii.1994, BGxh1, AN Recovery after 1993 fire, Pitfall trap No. C4 (G. G. E. Scudder) [SEM]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, NV, and UT (Bohart 1980)

Hyponysson bicolor Rohwer

1♀, Kamloops, 23.vii.1939 (G. J. Spencer) [SEM]; 2♀, Vancouver I., Saanich, Mt. Newton FC orchard, 16.vii–23.vii.1993, malaise (D. Blades) [PMAE]; 1♀, MacGillivray Cr. Game Reserve nr. Chilliwack, no date (no collector) [CNC]

Distribution: Genus and species new to Canada. Previously recorded from WA, ID, OR, and CA (Krombein 1979).

Microbembex nigrifrons (Provancher)

1 spm, Nicola, 3.viii.1923 (E. R. Buckell) [CNC]; 12 spms, Oliver, 22.viii.1923 (C. B. Garrett) [CNC]; 1♀, Osoyoos, 17.viii.1925 (G. J. Spencer) [SEM]; 1♂, Kamloops, 6.viii.1939 (G. J. Spencer) [SEM]; 1♂, Kamloops, 15.vii.1947 (B. A. Sugden) [RBCM]; 1 spm, Osoyoos, 25.viii.1953 (J. E. H. Martin) [CNC]; 1♂, Ashcroft, Rattlesnake Hills, bluff above Thompson R., 13.vi.1995, sand dunes, 1750' (S. G. Cannings & L. R. Ramsey) [RBCM]; 1♀, Kamloops, Valleyview, 14.vi.1995, silt benches, sage/bunchgrass, 430 m (L. R. Ramsey) [RBCM]; 1♀, 6.4 km S of Gang Ranch Jct., 21.vi.1995, sand bluff, 520m (S. G. Cannings) [RBCM]; 1 spm, Osoyoos, E side of Osoyoos Lake, Inkameep Indian Reservation, 30.vi.1996, along sandy road (S. G. Cannings) [RBCM]; 1 spm, Osoyoos, E side, 22.vii.1997 (R. J. Cannings) [RBCM]

Distribution: Species new to Canada. Previously recorded from the U.S. and Mexico west of the 100th meridian (Krombein 1979).

Nysson chumash Pate

1♀, Mission City, 24.vii.1953 (W. R. M. Mason) [CNC]; 1♂, Robson, 20.viii.1963 (H. R. Foxlee) [SEM]; 1♂1♀, Osoyoos, Mt. Kobau, 8.vii–13.vii.1991, 990m (D. Blades & C. Maier) [RBCM]

Distribution: Species new to Canada. Previously recorded from CA (Krombein 1979).

Nysson fidelis Cresson

1♀, Nelson, 5.viii.1916 (F. W. L. Sladen) [CNC]; 1♀, Minnie L., 26.vii.1925 (E. R. Buckell) [CNC]; 1♂, Robson, 25.vi.1952 (H. R. Foxlee) [CNC]; 1♀, Gagnon Rd. 6 mi W Terrace, 28.vi.1960 (J. G. Chillcott) [CNC]; 1♂, Robson, 5.vii.1965 (H. R. Foxlee) [SEM]; 1♀, Duffy Lake Rd., 50.385086°N 122.366734°W, 17.vii.2014, nr. lake, 1132m (D. G. Holden) [PMAE]; 1♂, Osoyoos, Mt. Kobau, 4.vi–8.vii.1991, 990m (D. Blades & C. Maier) [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, MT, OR, CO, and WI (Krombein 1979).

Nysson neorusticus Bohart

1♀, Nelson, 5.viii.1916 (Sladen) [CNC]; 1 spm, Kaslo, 3.viii.1916 (Sladen) [CNC]; 1♂, Comox, Pt. Holmes, 5.vii.1933 (J. McDunnough) [CNC]; 1♂, MacGillivray Creek Game Reserve nr. Chilliwack, 14.vi.1953 (W. R. M. Mason) [CNC]; 1 spm, MacGillivray Creek Game Reserve nr. Chilliwack, 14.vi.1953 (G. J. Spencer) [CNC]; 41♂ 3♀, MacGillivray Creek Game Reserve nr. Chilliwack, 15.vi.1953 (S. D. Hicks / W. R. M. Mason / G. J. Spencer) [CNC]; 1♂, Ruskin, 26.vi.1926 (W. R. M. Mason) [CNC]; 2♂, Mission City, 4.vii.1953 (W. R. M. Mason) [CNC]; 2♀,

MacGillivray Creek Game Reserve nr. Chilliwack, 23.vii.1953 (W. R. M. Mason) [CNC]; 1♀, Horsethief Creek, 22.vi.1956 (J. Grant collection) [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, OR, CA, NV, UT, and WY (Krombein 1979).

Nysson plagiatu Cresson

1♂, Keremeos, 20.vi.1943 (E. R. Buckell) [SEM]

Distribution: Species new to BC. Previously recorded from QC, ON, and in the U.S. east of the Rockies (Krombein 1979; Buck 2004).

Nysson recticornis Bradley

1♂, Penticton, West Bench, 5.vi.1983, rangeland/orchard edge, malaise trap (S. G. Cannings) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 14.vi–3.viii.1987, *Purshia/Aristida* steppe, pan trap (S. G. Cannings) [SEM] 20♂1♀, Osoyoos, Mt. Kobau, 8.vii–13.vii.1991, 990m (D. Blades & C. Maier) [RBCM]; 1♀, Osoyoos, Mt. Kobau, 14.vii–23.viii.1991, 560m [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, ID, and CA (Krombein 1979).

Nysson sp. Cresson

1♀, Robson, 6.ix.1962 (H. R. Foxlee) [SEM]

Distribution: Species new to Canada and potentially new to science. Further study is required.

Oryttus laminiferus (Fox)

1♀, Oliver, UBC Geology Camp, 20.vii.1989, pine/thicket edge, Malaise trap (S. G. Cannings) [SEM]

Distribution: Genus new to BC and species new to Canada. Previously recorded from WA, ID, OR, CA, NV, and UT (Bohart 1968b; Krombein 1979).

Oryttus mirandus (Fox)

1♀, Chopaka, 49°00'41.19" N 119°40'25.37" W, 18.vii.1996, 486m (G. G. E. Scudder) [RBCM]

Distribution: Genus new to BC and species new to Canada. Previously recorded from WA, CA, and NV (Bohart 1968b; Krombein 1979).

Steniolia tibialis Handlirsch

1♂, Goldstream, 20.vii.1902 (no collector) [CNC]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, and NV (Krombein 1979; Bohart and Gillaspay 1985).

Zanysson texanus fuscipes (Cresson)

2 spms, Penticton, 7.viii.1916 (Sladen) [CNC]; 1 spm, Keremeos, 21.vii.1917 (Sladen) [RBCM]; 13 spms, Vernon, 25.vii.1917 (Sladen) [CNC]; 1 spm, Walhachin, 17.vii.1918 (E. R. Buckell) [RBCM]; 1 spm, Fairview, 7.viii.1919 (E. R. Buckell) [RBCM]; 1♂, Vernon, 15.vii.1920 (N.L. Cutler) [CNC]; 1♂ 1♀, Walhachin, 9.vii.1944 (G. J. Spencer) [SEM]; 1♂, Kamloops, 3.viii.1944 (G. J. Spencer) [SEM]; 1♀, Kamloops, 16.vi.1945 (G. J. Spencer) [SEM]; 1♂, Vernon, 17.vii.1946, in garden soil (H. B. Leech) [SEM]; 1♂, Vernon, 5.viii.1947 (B. A. Sugden) [RBCM]; 1♂, Penticton, West Bench, 11.vii.1988, rose thicket/grassland boundary, Malaise trap (S. G. Cannings) [SEM]; 1♀, Lillooet, 50.684643°N 121.921613°W, 17.vii.2014, sandy site, 229m (D. G. Holden) [PMAE]

Distribution: Genus and species new to Canada. Previously recorded from the U.S. (Krombein 1979).

Crabronidae: Crabroninae

Crabro tenuis Fox

1♀, Kamloops, 9.vii.1939 (G. J. Spencer) [SEM]

Distribution: Species new to BC. Previously recorded from QC to AB, WA, UT, CO, MI, NJ, GA, OK, and WV (Bohart 1976; Krombein 1979; Buck 2004; Leclercq 2008).

Crossocerus chromatipus Pate

1♀, Victoria, Langford, Upper Thetis Lake, west of the north end of the lake, 629 Peacock Place, 10U 4645 53687, 7.viii.1997 (Gordon Green) [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, OR, CA, and NV (Krombein 1979; Leclercq 2000).

Crossocerus elongatulus (Vander Linden)

1♂, Vancouver, 8.viii.1960 (G. J. Spencer) [SEM]

Distribution: Species new to BC. Previously recorded from AB, ON, QC, YT, NS, and much of eastern North America west to WA and UT (Krombein 1979; Leclercq 2000).

Ectemnius alpheus Pate

1♀, Kamloops, 2.viii.1938 (G. J. Spencer) [SEM]

Distribution: Species new to Canada. Previously recorded from WA, UT, and CA (Krombein 1979; Leclercq 2007).

Ectemnius cephalotes (Olivier)

1♀, Vancouver, 18.viii.1988 (G.G.E. Scudder) [SEM]; 1♀, Vancouver, 31.vii.1991 (N. C. Scudder) [SEM]; 1♀, Vancouver, 1.viii.1991 (J. Scudder) [SEM]; 1♂, Vancouver, 49.2281, -123.0870, 16.viii.2010 (C. G. Ratzlaff) [SEM]; 1♀, Vancouver, 49.2281, -123.0870, 22.vi.2013 (C. G. Ratzlaff) [CGR]; 1♀, Vancouver, 49.2281, -123.0870, 11.vii.2013 (C. G. Ratzlaff) [CGR]; 1♀, Richmond, 49.1426, -123.1005, viii.2014, with calliphorid prey (J. E. & N. D. Schellenberg) [SEM]; 1♀, Vancouver, 49.2281, -123.0780, v.2015, in cell in stump with calliphorid remains (C. G. Ratzlaff) [CGR]; 1♀, Vancouver, UBC, Beaty Biodiversity Museum, 49.2632, -123.2502, 7.vii.2015 (C. G. Ratzlaff) [SEM]; 1♀, Vancouver, 49.2281, -123.0780, 10.viii.2015 (C. G. Ratzlaff) [CGR]; 1♀, Vancouver, 49.2281, -123.0780, 16.viii.2015 (C. G. Ratzlaff) [CGR]

Distribution: Introduced species new to BC. Previously recorded from ON, QC, CT, NY, NJ, PA, MD, IL, and Europe (Krombein 1979; Leclercq 2007).

Ectemnius maculosus (Gmelin)

1♀, Spillimacheen, 3.vii.1914 (F. W. L. Sladen) [CNC]; 1♀, Keremeos, 21.vii.1917 (Sladen) [CNC]; 2♂ 6♀, Vernon, 24-25.vii.1917 (Sladen) [CNC]; 1♂, Vernon, 23.vii.1920 (N. L. Cutler) [CNC]; 1♂, Salmon Arm, 28.vii.1925 (A. A. Dennys) [CNC]; 1♀, Agassiz, 30.viii.1926 (H. H. Ross) [CNC]

Distribution: Species new to BC. Previously recorded from most of Canada and U.S. east of the 100th meridian including MO, CO, WV, ME, CT, GA, NY, and PA (Krombein 1979; Buck 2004; Leclercq 2007).

Ectemnius rufifemur (Packard)

1 spm, Thompson River, 12.viii.1914 (T. Wilson) [CNC]; 1 spm, Keremeos, 31.vii.1917 (Sladen) [CNC]; 1♀, Walhachin, 11.vii.1918 (E. R. Buckell) [SEM]; 1 spm, Nicola, 3.viii.1923 (E. R. Buckell) [CNC]; 2♀, Kamloops, 10.vii.1938 (G. J. Spencer) [SEM]; 1♀, Kamloops, Lac du Bois, 28.viii.1954 (G. J. Spencer) [SEM]; 1♀, Wilmer, 21.viii.1982 (S. G. Cannings) [SEM]

Distribution: Species new to BC. Previously recorded from CO, FL, KA, MI, and in the Transition and Austral Zones east of the Rockies (Krombein 1979; Leclercq 2007).

Ectemnius spiniferus (Fox)

1 spm, Okanagan, 4.viii.1915 (no collector) [CNC]; 3 spms, Vernon, 25.vii.1925 (Sladen) [CNC]; 1♂, Vernon, 8.vii.1921 (M. H. Ruhmann) [CNC]; 1 spm, Vernon, 9.vii.1923 (M. H. Ruhmann) [CNC]; 1 spm, Lillooet, 27.v.1925 (E. R. Buckell) [CNC]; 1♀, Lillooet, Seton Lake,

28.vi.1926 (J. McDunnough) [CNC]; 1 spm, Grand Forks, 31.viii.1951 (D. A. Arnott) [CNC]; 1♀, Lillooet R., nr. John Sandy Cr., 9.vi.1981 (R. J. Cannings) [SEM]; 1♂, Penticton, West Bench, 6.vi.1988 (S. G. Cannings) [SEM]

Distribution: Species new to BC. Previously recorded from AB, WA, OR, CA, NV, CO, and AZ (Strickland 1947; Krombein 1979; Leclercq 2007).

Larropsis tenuicornis (Smith)

1 spm, Okanagan, 4.viii.1915 (no collector) [CNC]; 2♂ 1♀, Vernon, 25.vii.1917 (Sladen) [CNC]; 1♂ 1♀, S. Ver., 1.viii.1917 (no collector) [CNC]; 1 spm, S. Ver., 12.viii.1917 (no collector) [CNC]; 1♀, Vernon, 14.viii.1923 (D. G. Gillespie) [CNC]; 1♂, Vernon, 24.viii.1923 (D. G. Gillespie) [CNC]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, and NV (Bohart and Bohart 1966).

Miscophus sp.

2♂ 3♀, Osoyoos, Mt. Kobau, 24–28.viii.1991, 370m, SOCAP–OY51 (D. Blades & C. Maier) [RBCM]; 1♀, Osoyoos, Haynes Ecological Reserve, 22.viii–28.ix.2005, BGxh1, AN Recovery after 1993 fire, Pitfall trap (G. G. E. Scudder) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 25.vii–30.viii.2006, BGxh1, AN Recovery after 1993 fire, Pitfall trap (G. G. E. Scudder) [SEM]

Distribution: Species new to Canada and potentially new to science. Further study is required.

Oxybelus ventralis Fox

1♂, Vernon, 25.vii.1917 (Sladen) [CNC]; 1♀, Vernon, 22.vii.1920 (R. C. Treherne) [CNC]; 2♂, Vernon, 23.vii.1920 (N. L. Cutler) [CNC]; 1♂, Vernon, 27.vii.1920 (R. C. Treherne) [CNC]

Distribution: Species new to Canada. Previously recorded from WA, OR, CA, NV, WY, AZ, and Mexico (Bohart and Schlinger 1957).

Pisonopsis birkmanni Rohwer

1♀, Oliver, UBC Geology Camp, 27.vii.1990, hawthorn thicket edge, malaise trap (S. G. Cannings) [SEM]

Distribution: Genus and species new to Canada. Previously recorded from CA, TX, and Mexico (Krombein 1979).

Pisonopsis clypeata Fox

1♂ 3♀, Penticton, West Bench, 4–5.vi.1983, rangeland/orchard edge, malaise trap (S. G. Cannings) [SEM]; 3♂, Penticton, West Bench, 14.vi.1987, rangeland/orchard edge, malaise trap (S. G. Cannings) [SEM]

Distribution: Genus and species new to Canada. Previously recorded from CA, NV, and WY (Krombein 1979).

Plenoculus propinquus Fox

1♂, Robson, 8.vi.1945 (H. R. Foxlee) [CNC]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, NV, UT, CO, AZ, and NM (Williams 1960).

Solierella affinis (Rohwer)

1♀, Aspen Grove, 12 km S, 9.vii.1988 (S. G. Cannings) [SEM]

Distribution: Genus new to BC and species new to Canada. Previously recorded from ID, CA, WY, CO, and KS (Krombein 1979).

Solierella peckhami (Ashmead)

1♀, Tahitan, 10.viii.1986 (S. G. Cannings) [SEM] 1♀, Ashcroft, Cornwall Hills, 12.vii.1989, 5650 ft. (R. A. Cannings & C. S. Guppy) [RBCM]; 1♀, Nicola River, 1 km E of Petit Creek bridge, 2.vii.1988 (G. E. Hutchings) [RBCM]

Distribution: Genus and species new to BC. Previously recorded from ON and much of eastern North America west to ID and CA, as well as an adventive species in HI and the Marshall Islands (Krombein 1979; Buck 2004).

Solierella plenoculoides similis (Bridwell)

1♀, Kaslo, 3.viii.1916 (Sladen) [CNC]; 1♀, Squamish, Diamond Head Trail, 13.viii.1953 (G. J. Spencer) [CNC]; 1♂, Manning Prov. Pk., Eastgate, 22.vi–15.vii.1986 (S. G. Cannings) [SEM]; 1♂, Victoria, Ten-Mile Point, 14.vi.1987 (R. A. Cannings) [RBCM]; 1♂, Oliver, UBC Geological Camp, 29.vi.1996 (S. G. Cannings) [RBCM]; 1♀, Arrow Lakes, 6 km NW of Arrow Park Ferry, Mosquito Creek, 11 U 432330 5552803, 18.vii.2010 (J. Sharkey) [RBCM]

Distribution: Genus and species new to BC. The subspecies *similis* has been previously recorded from OR and CA. The nominate subspecies has been recorded from ON and the eastern U.S. west to CO, TX, and AZ (Krombein 1979; Buck 2004).

Solierella sp. (inermis-group)

1♀, Ashcroft, 1.5 km NE, 12.viii.1989, sage flat, 1450' (C. S. Guppy) [RBCM]

Distribution: Species group new to BC and Canada. Species in this group have been previously recorded from ID, CA, CO, AZ, NM, TX, much of the southeastern U.S., and Mexico (Krombein 1979; Bohart 1990).

Tachysphex amplus Fox

4♀, Kamloops, 6.vii.1944 (G. J. Spencer) [SEM]; 1♀, Kamloops, 27.vi.1949 (G. J. Spencer) [SEM]

Distribution: Species new to Canada. Previously recorded from WA, ID, MT, OR, CA, NV, UT, WY, CO, AZ, NM, TX, KS, and Mexico (Pulawski 1988).

Tachysphex hopi Pulawski

1♂, Oliver, Seacrest Hill, 20.v.1987 (R. A. Cannings) [RBCM]

Distribution: Species new to BC. Previously recorded from AB, WA, ID, MT, OR, CA, NV, UT, WY, CO, and AZ (Pulawski 1988).

Tachysphex mundus Fox

1 spm, Osoyoos, Haynes Ecological Reserve, "The Throne" area, 49°05'37"N 119°31'18"W, 27.vii.1988, 1200' (C. S. Guppy) [RBCM]

Distribution: Species new to BC. Previously recorded from AB, SK, MT, CO, AZ, NM, TX, NE, KS, much of the eastern U.S., and Mexico (Pulawski 1988).

Tachysphex verticalis Pulawski

1♀, Castlegar, 24.vii.1974 (W. R. M.) [RBCM]

Distribution: Species new to Canada. Previously recorded from ID, CA, NV, UT, AZ, and Mexico (Pulawski 1988).

Tachysphex williamsi Bohart

1♂, Penticton, West Bench, 26.v.1987 (R. A. Cannings) [RBCM]

Distribution: Species new to BC. Previously recorded from AB, SK, ID, MT, OR, CA, NV, UT, WY, CO, NM, AZ, ND, NE, and Mexico (Pulawski 1988).

Tachytes nevadensis Bohart

1 spm, Wallhachin, 12.vii.1918 (E. R. Buckell) [RBCM]; 1♂, Wallhachin, 17.vii.1918 (E. R. Buckell) [RBCM]

Distribution: Species new to Canada. Previously recorded from WA, ID, OR, CA, NV, and UT (Krombein 1979; Bohart 1994a).

Tachytes sayi Banks

1♀, Penticton, 7.ix.1919 (E. R. Buckell) [SEM]; 1♀, Summerland, 26.vii.1920 (no collector) [SEM]; 2♂, Kamloops, 26.vii.1942 (G. J. Spencer) [SEM]; 2♂, Kamloops, 9.vii.1944 (G. J. Spencer) [SEM]; 2♂ 1♀, Kamloops, 15–16.vii.1945 (G. J. Spencer) [SEM]; 6♀, Kamloops, Lac du Bois, 14.vii.1946 (G. J. Spencer) [SEM]; 2♂, Chase, 14.vii.1946 (G. J. Spencer) [SEM]; 1♀, Stump Lake, 24.viii.1947 (B. A. Sugden) [RBCM]; 1♂ 1♀, Kamloops, 17.vii.1954, in cop. (G. J. Spencer) [SEM]; 3♂, Kamloops, Lac du Bois, 18–24.vii.1954 (G. J. Spencer) [SEM]; 1♀, Penticton, West Bench, 18.vii.1986 (R. A. Cannings) [RBCM]; 2♂, Ashcroft, Rattlesnake Hills, 10 U 6247 56259, 13.vi.1995, sand dunes and bluffs above Thompson R., 533m (S. G. Cannings & L. R. Ramsay) [RBCM]; 2♂, Osoyoos, Osoyoos L., Inkameep Indian Reserve, 30.vi.1996 (R. J. Cannings) [RBCM]; 2♀, Osoyoos, Haynes Ecological Reserve, 9.vii–9.viii.1996, BGxh1, Recovery after 1993 fire, pitfall trap (G. G. E. Scudder) [SEM]; 1♀, Lillooet, Scotchman Rd. at Retasket Dr., 50.67669°N 121.9461°W, 17.vii.2014 (T. Kimoto) [PMAE]

Distribution: Species new to BC. Previously recorded from AB, WA, OR, CA, TX, NE, OK, and KS (Strickland 1947; Krombein 1979; Bohart 1994a).

Crabronidae: Pemphredoninae

Diodontus bidentatus Rohwer

1♂, Kamloops, 4.vii.1937 (G. J. Spencer) [SEM]; 1♂, Kamloops, 10.viii.1954 (G. J. Spencer) [SEM]; 1♂, Dog Creek, 18.vi.1963 (G. J. Spencer) [SEM]; 1♂, Penticton, West Bench, 23.vi.1983 (S. G. Cannings) [SEM]; 1♀, Kelowna, Knox Mt., nr. Base, 8.vi.1988, silt cliff (S. G. Cannings) [SEM]; 1♂, Kinbasket Reservoir, Canoe Reach (near Valemount), 354086 5848537, 12.vi.2010, 755m (N. Shaw & H. Schindler) [RBCM]; 1♀, Kinbasket Reservoir, Canoe Reach (near Valemount), 361065 5841696, 21.vi.2010, 752m (N. Shaw & H. Schindler) [RBCM]; 1♀, Kinbasket Reservoir, Canoe Reach (near Valemount), 354017 5848664, 12.vi.2010, 756m (N. Shaw & H. Schindler) [RBCM]; 1♀, Kinbasket Reservoir, Bush Arm (approx. 60 km NNW of Golden), 472087 5736588, 10.vii.2010, 755m (N. Shaw & H. Schindler) [RBCM]

Distribution: Species new to BC. Previously recorded from AB, QC, NB, AK, ID, MT, CO, NB, NY, MI, ND, and PA (Krombein 1979; Eighme 1989; Buck 2004)

Diodontus crassicornus Viereck

1♂, Penticton, West Bench, 8.vi.1988 (S. G. Cannings) [SEM]

Distribution: Species new to Canada. Previously recorded from WA, OR, CA, NV, UT, WY, CO, AZ, NM, AL, IA, MO, and WI (Eighme 1989).

Diodontus flavitarsis Fox

1♀, Departure Bay, 22.v.1925 (G. J. Spencer) [SEM]; 5♀, Chilcotin, 23.vi.1929 (G. J. Spencer) [SEM]; 3♂, Chilcotin, 16.vi.1930 (G. J. Spencer) [SEM]; 1♂ 1♀, Chilcotin, 23.vi.1930 (G. J. Spencer) [SEM]; 1♀, Nicola, 10.vii.1932 (G. J. Spencer) [SEM]; 1♀, Kamloops, 24.vi.1936 (G. J. Spencer) [SEM]; 1♀, Upper Clearwater, Edgewood, 25.vi.1989, Malaise (S. G. Cannings) [SEM]; 1♀, Kinbasket Reservoir, Canoe Reach (near Valemount), 474550 5738934, 10.vii.2010, 748m (N. Shaw & H. Schindler) [RBCM]

Distribution: Species new to BC. Previously recorded from YT, WA, ID, OR, CA, UT, WY, CO, AZ, NM, TX, DC, IA, MO, and PA (Eighme 1989; Buck 2004).

Diodontus fraternus Rohwer

1♂, Oliver, UBC Geology Camp, 20.vii.1989, pine/thicket edge, Malaise trap (S. G. Cannings) [SEM]; 1♀, Penticton, West Bench, 14.vi.1987, grassland/orchard edge, Malaise trap (S. G. Cannings) [SEM]

Distribution: Species new to Canada. Previously recorded from ID, CA, NV, UT, CO, AZ, NM, NE, and IA (Eighme 1989).

Diodontus occidentalis Fox

1♂, Oliver, UBC Geology Camp, 23.vii.1989, pine/thicket edge, Malaise trap (S. G. Cannings) [SEM]

Distribution: Species new to Canada. Previously recorded from AK, ID, CA, NV, UT, WY, CO, AZ, MI, NY, and ND (Eighme 1989).

Diodontus virginianus Rohwer

1♂, Penticton, West Bench, 23.viii.1987 (S. G. Cannings) [SEM]

Distribution: Species new to BC. Previously recorded from ON, ID, OR, CA, NY, MD, VA, and DC (Eighme 1989; Buck 2004).

Mimesa dawsoni Mickel

3♂, Rattlesnake Hills, 10U 6252 56232, 13.vi.1995, sand dunes and bluffs above Thompson River, 533m (S. G. Cannings) [RBCM]

Distribution: Species new to BC. Previously recorded from AB, MB, ON, MT, UT, KS, OK, ND, SD, NC, NJ, NY, MI, MA, IL, IA, MN, and NB (Finnamore 1983).

Mimesa sabina Gittins

1♂, Vernon, 25.vii.1917 (Sladen) [CNC]

Distribution: Species new to Canada. Previously recorded from CA, NV, UT, and CO (Finnamore 1983).

Mimusesa clypeata (Fox)

1♀, Invermere, 18.vii.1928 (A. A. Dennys) [RBCM]; 1♂, Chilcotin, 12.vii.1930 (G. J. Spencer) [SEM]; 3♀, Blind Bay, vi.1987 (C. A. Elsey) [PMAE]; 3♀, Blind Bay, 15.vii.1987 (C. A. Elsey) [PMAE]

Distribution: Species new to BC. Previously recorded from AB, YT, NT, SK, NL, AK, ID, WA, CA, NV, UT, and CO (Krombein 1979; Buck 2004).

Psenulus frontalis (Fox)

1 spm, Lillooet, Seton Lake, 3.vi.1926 (J. McDunnough) [CNC]; 1 spm, Robson, 28.vi.1950 (H. R. Foxlee) [CNC]; 2 spms, Summerland, Gartrell Point, 49°34'22"N 119°37'06"W, 29.vi.1990, 1140', Malaise (R. A. Cannings and H. Nadel) [RBCM]; 1♂ 6♀, Blind Bay, vii.1987, malaise (C. A. Elsey) [PMAE]

Distribution: Species new to Canada. Previously recorded from WA, CA, UT, CO, AZ, and NM (Krombein 1979).

Stigmus fraternus Say

1♀, Cranbrook, 21.vi.1926 (A. A. Dennys) [RBCM]; 1 spm, Robson, 16.viii.1950 (H. R. Foxlee) [CNC]; 2 spm, Alaska Hwy., Mi. 496, Liard Hot Springs, 9–10.vii.1959, 1500' (R. E. Leech/E. E. MacDougall) [CNC]; 1♀, Osoyoos Lake, N end, v–vi.1985, *Betula/Alnus* woodland, pan trap (S. G. Cannings) [SEM]

Distribution: Species new to BC. Previously recorded from ON, QC, MT, and much of the eastern U.S. (Krombein 1973, 1979; Buck 2004).

Crabronidae: Philanthinae

Cerceris calochorti Rohwer

1♀, Osoyoos L., East side, 16.v.1993, sandy road (S. G. Cannings) [RBCM]

Distribution: Species new to BC. Previously recorded from southern Canada and the U.S. west to the eastern slopes of the Rockies including AB, MT, UT, WY, CO, NM, TX, ND, and SD (Scullen 1965; Krombein 1979).

Philanthus bilunatus Cresson

1♂, Quesnel, 15.vii.1948, on mustard (H. R. MacCarthy) [SEM]; 1♀, Soda Creek, 29.vii.1945 (H. R. MacCarthy) [SEM]; 1♀, Quesnel, 11.viii.1948 (G. J. Spencer) [SEM]; 2♂ 1♀, Soda Creek, 9.vii.1950 (G. J. Spencer) [SEM]; 2♀, Cottonwood R., viii.1953 (J. Grant) [RBCM]; 1♂, Kleena Kleene, Kliniklini River, East crossing, 29.vii.1987 (R. A. Cannings and R. J. Hebda) [RBCM]

Distribution: Species new to BC. Previously recorded from NS to AB, PE and the eastern U.S. west to ND, CO, and NM (Bohart and Grissell 1975; Buck 2004).

Sphecidae: Ammophilinae

Ammophila acuta (Fernald)

1♀, Fairmont Hot Springs, Dutch Cr., 1.vii.1982 (G. G. E. Scudder) [SEM]; 1♂, Penticton, West Bench, 18.vi.1989, orchard/grassland edge, Malaise trap (S.G. Cannings) [SEM]; 1♂, Oliver, UBC Geology Camp, 23.vii.1989, pine/thicket edge, Malaise trap (S. G. Cannings) [SEM]; 2♀, Summerland, Agriculture Canada Research Station, grassland research site, 25.vi.1990 (S. G. Cannings) [SEM]; 1♂, Windermere Valley, 50°21.47' N 115°55.10' W, 26.vii–17.ix.2000, Pitfall trap #5 (R. Sargent) [SEM]

Distribution: Species new to Canada. Previously recorded from the western U.S. (Menke 1965).

Ammophila ferruginosa Cresson

1♂, Osoyoos, H.E.R., 29.v.1987 (S. G. Cannings) [SEM]

Distribution: Species new to BC. Previously recorded from AB, MB, ID, CA, NV, UT, WY, CO, AZ, NM, TX, KS, NE, OK, ND, MN, and Mexico (Menke 1965).

Ammophila karenae Menke

1♂, Ashcroft, 1 km N, 7.viii.1982 (R. A. Cannings) [RBCM]; 1♀, Osoyoos, H.E.R., 20.vi–10.vii.1986, sand/*Artemisia/Purshia*, pitfall (S. G. Cannings) [SEM]; 1 spm, Lillooet Dist., no date (no collector) [RBCM]

Distribution: Species new to Canada. Previously recorded from ID, CA, NV, AZ, NM, and Mexico (Krombein 1979).

Ammophila mcclayi Menke

2♀, Chopaka, 21.v.1986, sage flats (R. A. Cannings) [RBCM]

Distribution: Species new to Canada. Previously recorded from CA, NV, and NM (Menke 1965; Krombein 1979)

Podalonia clypeata Murray

1♀, Vaseux L., 4.viii.1984 (S. G. Cannings) [SEM]

Distribution: Species to Canada. Previously recorded from WA, OR, MT, NM, KS, OK, and MN (Murray 1940).

Podalonia occidentalis Murray

1♀, Shuswap Narrows, 31.viii.1943 (G. J. Spencer) [SEM]; 1♂, Osoyoos, Haynes Ecological Reserve, 20.vi.1986 (S. G. Cannings) [SEM]; 1♀, Oliver, IR1, 'Water tower', 49°10' N 119°31' W, 1.vi–7.vii.1994, BGxh, *Purshia* assoc., Pitfall trap U2-1 (G. G. E. Scudder) [SEM]

Distribution: Species new to BC. Previously recorded from AB, WA, ID, MT, OR, CA, UT, WY, CO, AZ, and NM (Murray 1940; Krombein 1979).

Sphecidae: Chloriontinae

Chlorion aerarium Patton

1♀, Oliver, 27.viii.1957 (J. Boone) [SEM]

Distribution: Species new to BC. Previously recorded from ON and the entire U.S. (Krombein 1979; Buck 2004).

Doubtful Records

Two species, *Ammophila conditor* Smith and *Cerceris deserta* Say, were recorded from Victoria on Vancouver Island by Taylor in 1884. *Ammophila conditor* Smith has only elsewhere been recorded from Florida; *C. deserta* Say has only been recorded from northeastern North

America east of the Rockies. These specimens are almost certainly misidentified due to the confused taxonomy of these genera, which was not cleared up for another 71 years (Menke (1965) for *Ammophila* and Scullen (1965) for *Cerceris*). Both records were also ignored by the authors of their respective genera revisions.

Spencer and Wellington wrongly recorded the species *Chlorion aztecum* de Saussure, a synonym of *Isodontia azteca* (de Saussure), from Summerland in 1948. *Isodontia azteca* (de Saussure) is a Neotropical species with a range that extends only to southeastern U.S.

Also not included was the species *Podalonia argentipilis* (Provancher). Originally described from Vancouver in 1887, the holotype was recorded as lost by Gahan and Rohwer (1918). Krombein (1958) synonymized *Podalonia morrisoni* Cameron, a species that was known only from southern California, Southern Arizona, and Mexico, with *P. argentipilis* (Provancher), known only from that single specimen. If this synonymy is correct, then Provancher's type specimen was likely mislabelled and originally came from the southwestern United States. If incorrect, then the specimen was from British Columbia, but the identity cannot be verified and has therefore not been included.

Provincial Distribution

Spheciform wasps are found in all habitats throughout the province, from alpine/subalpine areas to lush valley bottoms. Many genera, however, prefer hot, dry habitats with sand or sandy soil for nesting and are, therefore, limited in their distribution. The Southern Interior Ecoprovince, including the Okanagan–Similkameen region of British Columbia, contains Canada's only true desert and thus the greatest diversity of spheciform wasps. Of the 280 spheciform wasp species found in British Columbia, 77 (27.5% of total species) are limited to the Southern Interior in their distribution; of these, 51 (18.2% of total species) are found nowhere else in Canada. This area represents the northern-most extent of the range for many species, with almost all extending down through Great Basin ecosystem in the United States.

National Distribution

Six provincial checklists of spheciform wasps have been produced in Canada, with three focusing specifically on the three Canadian families and three serving as parts of a more extensive checklist of provincial Hymenoptera. Table 2 summarizes the number of species for each subfamily record in each checklist. The numbers present are based exclusively on the checklists, adjusted for synonymies, and do not take into account newer records that are almost certainly present for some provinces.

Ontario has the highest number of recorded species at 284; British Columbia has the second-highest number at 280. Both provinces have a large number of species found nowhere else in Canada. The high diversity is likely due to the presence of the northern-most parts of warmer southern ecosystems. In British Columbia, this is the Great Basin ecosystem, which reaches the Southern Interior. In Ontario, the Carolinian life zone just reaches into southwestern part of the province (Buck 2004).

Introduced Species

Two species of crabronid wasps have been introduced to British Columbia in the past 35 years. *Ectemnius cephalotes* (Olivier) and *Passaloecus singularis* Dahlbom, both native to Europe and Western Asia, have been recorded from the Lower Mainland (Georgia Depression) region of British Columbia. Both were initially recorded in North America on the east coast and have expanded throughout multiple states and provinces. It is still uncertain whether the eastern population of *E. cephalotes* (Olivier) is, in fact, introduced or if it is simply a disjunct population, since Provancher described a synonymous species from Quebec in 1882. In Canada, *E. cephalotes* (Olivier) has previously been recorded only from Ontario and Quebec, whereas *P. singularis* Dahlbom has been recorded from British Columbia, Ontario, and Quebec (Vincent 1978; Buck 2004). Both species nest in wooden cavities, such as beetle borings in dead wood—a characteristic that would allow for human-facilitated travel over large distances (Warburton 1920; Vincent 1978).

Table 2

Species totals by subfamily for the six published provincial checklists containing spheciform wasps.

Provincial Checklist	Total # of Species	Ampulicidae			Sphecidae					Crabronidae				Unique Species
		Amp	Dol	Amm	Chl	Sce	Sph	Ast	Bem	Cra	Mel	Pem	Phi	
BC (Ratzlaff 2015)	280	0	0	27	1	2	9	16	47	95	0	55	28	122
AB (Strickland 1947*)	121	0	0	20	0	2	3	3	15	42	1	19	16	22
ON (Buck 2004/2005)	284	1	1	15	1	3	11	7	40	117	2	56	30	93
QC (Finnamore 1982)	155	0	0	11	0	2	4	4	21	66	1	31	15	15
YT (Finnamore 1997*)	68	0	0	4	0	0	0	7	6	29	0	21	1	3
NWT (Steiner 1973*)	34	0	0	5	0	0	0	3	5	14	0	6	1	0

Subfamilies are abbreviated as follows: Amp – Ampulicinae, Dol – Dolichurinae, Amm – Ammophilinae, Chl – Chloriontinae, Sce – Sceliphrinae, Sph – Sphecinae, Ast – Astatinae, Bem – Bembicinae, Cra – Crabroninae, Mel – Mellininae, Pem – Pemphredoninae, Phi – Philanthinae. Unique species refers to the number of species recorded, in Canada, from only that province. * checklist of spheciform wasps as part of larger study of provincial Hymenoptera

Identification Resources

The following table summarizes the primary available resources for the identification of spheciform wasps in British Columbia. Although limited to only the genera found in the province, it can still be useful for surrounding areas since almost all the papers listed include all North American species.

CONCLUSION

The amazing diversity in morphology, habitats, and biology of spheciform wasps in this environmentally heterogeneous province is fitting. There is likely much more to be discovered. The Southern Boreal Interior Ecoprovince is by far the most understudied area in British Columbia when it comes to spheciform wasps. Even light studies that focused on these wasps would likely expand the distributions of many species, increasing the total numbers for the ecoprovince. Another area in need of study is the northeastern corner of British Columbia, which contains the Boreal and Taiga Plains ecoprovinces. Little sampling has been done, and potential exists for detection of transcontinental boreal species and central North American species due to habitat that is unique in British Columbia but similar to that found in northern parts of the Canadian prairies.

This checklist also highlights the importance of the Southern Interior to biodiversity in British Columbia. Many species found in the region are found nowhere else in the province or country, further emphasizing the need for the protection of the habitats in which these species live. Because many of the specimens examined were collected prior to 1990, sampling is needed to determine the current status of species in the area. As well, little is known about the biology of many of these wasps, and future research of spheciform wasps in British Columbia has much potential.

Table 3

Bibliography of available resources for the identification of British Columbian spheciform wasps.

Genus	References for Identification	Genus	References for Identification
Crabronidae: Astatinae		Crabronidae: Pemphredoninae	
<i>Astata</i>	Parker (1962)	<i>Ammoplanus</i>	Smith (2008)
<i>Diploplectron</i>	Parker (1972)	<i>Diodontus</i>	Eighme (1989)
<i>Dryudella</i>	Parker (1969)	<i>Mimesa</i>	Finnamore (1983)
Crabronidae: Bembicinae		<i>Parammoplanus</i>	Smith (2010)
<i>Alysson</i>	Fox (1894b)	<i>Passaloecus</i>	Vincent (1978)
<i>Bembix</i>	Bohart and Horning (1971)	<i>Pemphredon</i>	Dollfuss (1995)
<i>Bicyrtes</i>	Bohart and Horning (1971)	<i>Psenulus</i>	N/A
<i>Clitemnestra</i>	N/A	<i>Pulverro</i>	Smith (1983)
<i>Didineis</i>	Malloch and Rohwer (1930)	<i>Spilomena</i>	Bohart and Smith (1995)
<i>Epinysson</i>	N/A	<i>Stigmus</i>	Krombein (1973)
<i>Glenostictia</i>	Bohart and Gillaspys (1985)	Crabronidae: Philanthinae	
<i>Gorytes</i>	N/A	<i>Aphilanthops</i>	Bohart and Grissell (1975)
<i>Harpactus</i>	Bohart (1980)	<i>Cerceris</i>	Scullen (1965); Bohart and Grissell (1975); Ferguson (1984)
<i>Hoplisoides</i>	Bohart (1997); Buck (2007)	<i>Clypeadon</i>	Bohart and Grissell (1975)
<i>Hyponysson</i>	N/A	<i>Eucerceris</i>	Scullen (1968); Bohart and Grissell (1975)
<i>Lestiphorus</i>	N/A	<i>Philanthus</i>	Bohart and Grissell (1975)
<i>Microbembex</i>	Bohart and Horning (1971)	Sphecidae: Ammophilinae	
<i>Nysson</i>	N/A	<i>Ammophila</i>	Menke (1965)
<i>Orytus</i>	Bohart (1968b)	<i>Podalonia</i>	Murray (1940)
<i>Seniolia</i>	Bohart and Gillaspys (1985)	Sphecidae: Chloriontinae	
<i>Stictiella</i>	Bohart and Gillaspys (1985)	<i>Chlorion</i>	N/A
<i>Stizoides</i>	Gillaspys (1963)	Sphecidae: Sceliphrinae	
<i>Zanysson</i>	Rohwer (1921)	<i>Chalybion</i>	Hensen (1988)
Crabronidae: Crabroninae		<i>Sceliphron</i>	van der Vecht and van Bruegel (1968)
<i>Belomicrus</i>	Bohart (1994b)	Sphecidae: Sphecinae	
<i>Crabro</i>	Bohart (1976); Leclercq (2008)	<i>Isodontia</i>	Bohart and Menke (1963)
<i>Crossocerus</i>	Leclercq (2000)	<i>Palmodes</i>	Bohart and Menke (1961)
<i>Ectemnius</i>	Bohart and Kimsey (1979); Leclercq (2007)	<i>Prionyx</i>	Parker (1960); Bohart and Menke (1963)
<i>Larropsis</i>	Bohart and Bohart (1962); Bohart and Bohart (1966)	<i>Sphex</i>	Bohart and Menke (1963)
<i>Lestica</i>	Leclercq (2006)		
<i>Lindenius</i>	N/A		
<i>Liris</i>	Krombein and Shanks Gingras (1984)		
<i>Lyroda</i>	N/A		
<i>Miscophus</i>	N/A		
<i>Oxybelus</i>	Bohart and Schlinger (1957)		
<i>Pisonopsis</i>	Williams (1954)		
<i>Plenoculus</i>	Williams (1960)		
<i>Rhopalum</i>	Leclercq (2002)		
<i>Solierella</i>	Williams (1950); Bohart (1990)		
<i>Tachysphex</i>	Pulawski (1988)		
<i>Tachytes</i>	Bohart (1994a)		
<i>Trypoxylon</i>	Sandhouse (1940)		

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Assessing a method for rearing North American yellowjackets

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ABSTRACT

Studying yellowjackets is challenging due to their cryptic nesting behaviour, short field season, and extreme variation in population density. Developing or perfecting techniques for rearing yellowjackets would greatly increase the opportunity of studying the communication ecology of yellowjackets and the evolution of eusociality in the Hymenoptera. Our objective was to assess a method for rearing the five *Vespula* congeners *V. acadica* (Sladen), *V. alascensis* (Packard), *V. atropilosa* (Sladen), *V. germanica* (F.), and *V. pensylvanica* (Saussure). In early spring 2014, we collected queens of each of the five species from the field and placed them singly in a plywood nest box connected to a mesh cylinder that served as a foraging arena and provided constant access to water and food (honey, live flies, and live caterpillars). For each queen, we recorded nest initiation, the attachment site of the nest pedicel, and the stage of nest development at the end of the experiment, nine weeks after the last collection date of queens. Queens of *V. germanica* (n=18), *V. alascensis* (n=11), *V. acadica* (n=4), *V. pensylvanica* (n=23), and *V. atropilosa* (n=11) had nest-initiation rates of 61%, 50%, 25%, 17%, and 0%, respectively. The mean number of nest cells built by queens of *V. germanica*, *V. alascensis*, *V. acadica*, *V. pensylvanica*, and *V. atropilosa* were 21.6 ± 4.6 , 17.8 ± 6.3 , 8.0 , 26.5 ± 8.3 , and 0 , respectively. Two *V. germanica* queens and one *V. pensylvanica* queens established nests that produced a few worker wasps. Although our rearing method compares favorably to, and in some aspects improves, previous rearing methods, further refinements are needed to generate the large numbers of wasp workers that are essential for experimental testing of hypotheses pertinent to life-history traits of yellowjackets.

Key Words: Yellowjacket, *Vespula*, rearing, nesting

INTRODUCTION

Yellowjackets and hornets are intensely studied because they can be (i) invasive and pestiferous species in many ecosystems (Landolt 1998; D'Adamo *et al.* 2001; Day and Jeanne 2001; Landolt *et al.* 2005; Brown *et al.* 2014), (ii) potential biological control agents (Hoffmann *et al.* 2000), (iii) vectors of microorganisms (Davis *et al.* 2012; Stefanini *et al.* 2012), and (iv) threats to citizens with venom (hyper)sensitivity (Nakajima 1986; Schmidt 1986; Ono *et al.* 2003). Furthermore, wasps are model organisms for studying the evolution of eusociality (Landolt *et al.* 1998) and chemotaxonomy (Bruschini *et al.* 2007). However, studies of wasps are challenging due to a short field season, extreme variation in wasp-population densities, and the often cryptic nesting behaviour of wasps (Edwards 1980).

There are seven accounts of establishing vespine nests in the laboratory (Table 1). Ishay *et al.* (1967) reared *Vespa orientalis* L. with field-collected nests and overwintered gynes in their "Vespiaries", but did not comment on the success rate of either method. Ross *et al.* (1981) attempted to rear nests of five *Vespula* species under laboratory conditions, and recorded the percent of nest initiation for each of these species. None of the nests developed beyond the emergence of the first workers. Following up on the work by Ross *et al.* (1981), Matthews *et al.* (1982) reared 14 nests of *V. maculifrons* (Buysson), five nests of *V. germanica* (F.), one nest of *V. vulgaris* (L.) [= *V. alascensis* (Packard)], and one nest of *V. vidua* (Saussure) under environmentally controlled conditions. All of these nests progressed to producing at least two queen larvae. Using the method by Ross *et al.* (1981), Ross (1983) reared and studied queen foraging behaviour of *V. germanica*, *V. vulgaris* (= *V. alascensis*), and *V. maculifrons*, using 3–5 nests of each species. Vetter and Visscher (1995) successfully reared nests of *V. pensylvanica* (Saussure) from field-collected gynes, reporting the first and only account of laboratory-reared

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Table 1
Description of methodologies and success rates for rearing yellowjackets and hornets in the genera *Vespa* and *Vespula* (V.)

Species (# gynes not specified)	Collection dates	Gynes nest-building	Nests producing Workers'	Gynes/males produced	Overwint. generation	Size (cm) of nest box	Diet	Rearing condition	Ref.
<i>Vespa orientalis</i> ; numbers not specified	N/A:	Yes; numbers not specified	Yes; numbers not specified	N/A	Yes	55×55×10 55×55×5	Free forage; bee hive	Indoor shed; near-outdoor conditions	1
<i>V. germanica</i> (22 OW; 21 FC)	May-June 1980;	28% (12/43)	42% (5/12)	No	No	1.1–0.6-L paper cylinder lined with soil & root surrogate	Water; honey; live flies; frozen crickets; cat food; frozen fish; hamburgers	Photoperiod: L: 15h, 20min D: 8h, 40min Temp: L: 24±2°C D: 18±2°C RH: 65–90%	2
<i>V. maculifrons</i> (54 FC)	Overwint. gynes from fall 1979	48% (26/54)	15% (4/26)						
<i>V. alascensis</i> (11 FC)		27% (3/11)	33% (1/3)						
<i>V. flavopilosa</i> (14 FC)		0% (0/14)	(0/0)						
<i>V. vidua</i> (7 FC)		28% (2/7)	50% (1/2)						
<i>V. germanica</i> (N/A:)	May-June 1980	5	Yes; numbers unspecified	Yes	No	As in Ref 2	As in Ref 2	As in Ref 2	3
<i>V. maculifrons</i> (N/A:)		14							
<i>V. alascensis</i> (N/A:)		1							
<i>V. vidua</i> (N/A:)		1							
<i>V. alascensis</i> (N/A:)	April- May 1983	3	N/A:	No	No	As in Ref 2	As in Ref 2	As in Ref 2	4
<i>V. maculifrons</i> (N/A:)		5							
<i>V. germanica</i> (N/A:)		5							
<i>V. pensylvanica</i> (24)	March-June 1994	46% (11/24)	45% (5/11)	2/4 produced gynes;	No	0.4-L cylinder lined with paper; root surrogate on lid	Honey/water; cat food; chicken; bee larvae/pupae; salmon; crickets; cabbage looper larvae	Temp.: 25°C Photoperiod: as outdoors	5
<i>V. vulgaris</i> (11)	Springs of 1994-95	45% (5/11)	40% (2/5)	2/4 produced males	No	15×10×10; soil-lined	Honey/water; live blowflies	Indoors with near-outdoor conditions	6
<i>V. germanica</i> (5 OW; 33 FC)		29% (11/38)	9% (1/11)	No	No				
<i>Vespa crabro</i> (5)	October 1998	20% (1/5)	1/1 nest produced workers	12 males; 3 gynes	Yes	30×20×40	Bee drones & pupae	Temp: 20-30°C; RH:70%	7
<i>V. alascensis</i> (10)	March-May 2014	50% (5/10)	20% (1/5)	No	No	15×15×30	Honey/ syrup; house flies; cabbage looper larvae	Natural temp., RH & light	8
<i>V. acadica</i> (4)		25% (1/4)	25% (1/4)						
<i>V. germanica</i> (18)		61% (11/18)							
<i>V. atropilosa</i> (11)		0% (0/11)							
<i>V. pensylvanica</i> (23)		17% (4/23)							

1OW = Overwintered; FC = Field collected; 2N/A = No information provided; 3Subset of those queens that initiated nests; 4(1) Ishay et al. 1967; (2) Ross et al. 1981; (3) Matthews et al. 1982; (4) Ross 1983; (5) Vetter and Visscher 1995; (6) Leathwick 1997; (7) Hoffmann et al. 2000; (8) this study

Vespula nests from spring-captured queens through to males and gynes. In New Zealand, Leathwick (1997) reared one *V. germanica* nest and two *V. vulgaris* nests, which produced workers. Finally, Hoffmann *et al.* (2000) mated gynes and males from two feral *Vespa crabro* L. nests and over-wintered five mated gynes, of which one established a nest that produced next-generation gynes.

Both transplanting feral nests into research areas and in situ observations and experimentation are means of studying eusocial wasps (Spradbery 1973; Edwards 1980; Akre *et al.* 1980; Akre 1982) and advancing our understanding of their ecology. However, the cryptic nesting behaviour of vespine wasps and extreme variation in population density make it difficult to locate sufficient numbers of nests for rigorous and replicated observations and experiments (Edwards 1980; Aldiss 1983). Furthermore, the process of excavating subterranean nests and transplanting them to new locations may damage the brood comb and nest envelope, thereby possibly affecting the behaviour of nest mates or leading to the loss of queens (Vetter and Visscher 1995).

A consistent supply of wasp nests would greatly benefit the study of vespine ecology, particularly the biology and ecology of the nest as a super-organism (Wilson 1971; Moritz and Bürgin 1987). This is most obvious in studies of alarm-pheromone systems among social wasps, where the presence of the nest is essential to observe nest-defense behavior. Of the nine species of yellowjackets and hornets that reportedly use alarm pheromones, pheromone components have been identified for only four species (Maschwitz 1964a,b; Saslavsky *et al.* 1973; Veith *et al.* 1984; Maschwitz 1984; Maschwitz and Hanel 1988; Heath and Landolt 1988; Landolt *et al.* 1995; Landolt *et al.* 1999; Ono *et al.* 2003), and the pheromone effect has often been tested with only a single nest.

Our objective was to assess a method for rearing *Vespula* congeners targeting for diversity *V. acadica* (Sladen), *V. alascensis*, *V. atropilosa* (Sladen), *V. germanica*, and *V. pensylvanica*.

MATERIALS AND METHODS

Collection of queens

We sweep-netted queens of unknown mating status in the Greater Vancouver area and Lillooet, both British Columbia (B.C.), during sunny clear days between 10:00 and 16:00 hours, capturing most queens while they were prey-hunting or collecting nectar from English hedge laurel, *Prunus lauresianus*. Between 20 March and 15 May 2014, we collected a total of 66 yellowjacket queens [*V. acadica* (4), *V. alascensis* (10), *V. atropilosa* (11), *V. germanica* (18), and *V. pensylvanica* (23; Table 2)], 55 of which in Vancouver, and 11 of which during a 2-day trip (13–15 May) to Lillooet. We immediately placed captured queens singly into glass jars (0.3–0.5 L) containing foliage of *P. lauresianus* or Western red-cedar, *Thuja plicata*, on which they commonly rest. Whenever possible, we kept jars in a cool and dark area for <2 h before placing them in rearing units (see below) that we kept inside a fenced area of Simon Fraser University's (SFU's) insectary annex. This approach minimized the queens' stress of confinement.

Rearing units

Nest-rearing units resembled those described by Ross *et al.* (1981), but had several modifications (Figure 1). Each unit consisted of a plywood box nesting cavity (15 cm high × 15 cm wide × 30 cm long) with one side panel hinged for periodic observations. A 2.5-cm hole in the top (dorsal) panel of the nest box provided entry into a mesh screen cylinder (15 cm diam × 20 cm tall), the top and bottom of which was hot-glued to a Petri dish (15 cm diam) for rigidity and stability. A hole (5 cm diam) in the bottom Petri dish of the cylinder corresponded with the dorsal hole of the nest box, allowing the foundress and potential workers to exit the box and to enter the mesh cylinder for foraging. The top Petri dish of the cylinder had one hole (3 cm diam) to accommodate an inverted 50-mL falcon tube with a cotton-filled pipette tip containing the water supply, and a second hole (2 cm diam) that was plugged with a cork or rubber stopper and allowed intermittent insertion of live flies and cabbage looper larvae (see below) as food sources. The top of the falcon tube was cut off to replenish water as it was consumed or evaporated (Figure 1).

Table 2

Numbers of overwintered *Vespula* (V) queens field-collected between 20 March and 15 May 2014, the proportion of queens that initiated nests and attached the nest pedicel to a twig in the nest box (Figure 1b) or the nest box roof, and the mean number of cells built by queens that initiated nests.

Species	# Queens collected	# Queens initiating nests*	Attachment site of pedicel in nest box ^a		Cells built ^a Mean (SE)
			Twig	Roof	
<i>V. acadica</i>	4	1/4 (25%)	1/1 (100%)	0/1 (0%)	8
<i>V. alascensis</i>	10	5/10 (50%)	3/5 (60%)	2/5 (40%)	17.8 (6.3)
<i>V. atropilosa</i>	11	0/11 (0%) **	-	-	-
<i>V. germanica</i>	18	11/18 (61%) **	5/11 (45%)	6/11 (55%)	21.6 (4.6)
<i>V. pensylvanica</i>	23	4/23 (17%)	3/4 (75%)	1/4 (25%)	26.5 (8.3)

^aSubset of those queens that initiated nests
* Significant difference between nest initiation rates (p=0.0013, FET)
** Significant difference in nest initiation rates between queens of *V. germanica* (61%) and *V. atropilosa* (0%; p=0.012, FET)

In previous studies for rearing yellowjackets, root surrogates as potential attachment sites for the nest pedicel were affixed to the top of nesting boxes (Ross *et al.* 1981; Matthews *et al.* 1982; Ross 1983, Vetter and Visscher 1995, Leathwick 1997). Accordingly, we hot-glued a few twigs as surrogate roots to the roof of nesting boxes, and tracked whether queens attached the nest pedicel to a twig or the roof of the box.

Some of the most vigorous nests of *V. alascensis* and *V. germanica* we observed in the field were built in straw composts, as previously reported (Spradbery 1971). Therefore, we lightly packed the 6.5-L nest box with (untreated) organic animal bedding straw for insulation. We supplied the mesh cylinder with decaying wood and filter paper to encourage pulp gathering for nesting material.

To prevent predation by ants, we placed each rearing unit on a brick in a water-filled tray on a table about 1 m above ground in a south-facing, rain-sheltered area.

Food provisioning of queens

Starting on the day of capture, we fed each queen daily with (i) honey and/or corn syrup (Akre *et al.* 1976; Ross *et al.* 1981) that we smeared on the mesh cylinder of the rearing unit, (ii) 3–5 common house flies, *Musca domestica* L., or the bottle flies *Lucilia sericata* (Meigen) or *Phormia regina* (Meigen) (Akre *et al.* 1976), and (iii) 5–10 2nd or 3rd instar larvae of the cabbage looper, *Trichoplusia ni* (Hübner), the latter also used as prey for yellowjackets by Vetter and Visscher (1995), although these were rarely consumed in our study. Although we often observed queens hunting flies, consuming honey and water, and entering or exiting the nesting box, we never observed violent flights against the mesh screen cylinder in attempts to escape the confinement. To reduce disturbance of nesting behaviour, we checked for nest initiation only once per week, always when the queen was foraging in the mesh cylinder.

Statistical analyses of data

We analysed all data with the statistical software R (version 3.1.3). We used a Pearson’s X² test, binomial exact test, or Fisher’s exact test (FET), depending on the data constraints and the specific hypothesis, to test for a difference in (i) nest-initiation rate between species (FET), followed by pairwise comparisons of proportions using the Bonferroni P-value adjustment method using the *fmsb* R package (Nakazawa 2014), (ii) site of pedicel attachment between species

(FET), and *iii*) for a deviation from a 50/50 chance of pedicel attachment to the twig or nest box roof for each species (X^2 goodness of fit test or binomial exact test), addressing the question whether queens have an innate preference for root-like substrates to attach the nest pedicel. To test for differences in the mean number of cells built by queens of the five species we studied, we performed an ANOVA.

RESULTS

Percent nest initiation

Queens of all five species, except *V. atropilosa*, initiated a nest. Queens of *V. germanica*, *V. alascensis*, *V. acadica*, and *V. pensylvanica* had nest-initiation rates of 61%, 50%, 25%, and 17%, respectively. The significant difference between these five nest-initiation rates ($p=0.0013$, FET; Table 1) can be attributed to significantly different nest initiation rates between queens of *V. germanica* (61%) and *V. atropilosa* (0%) ($p=0.012$, FET).

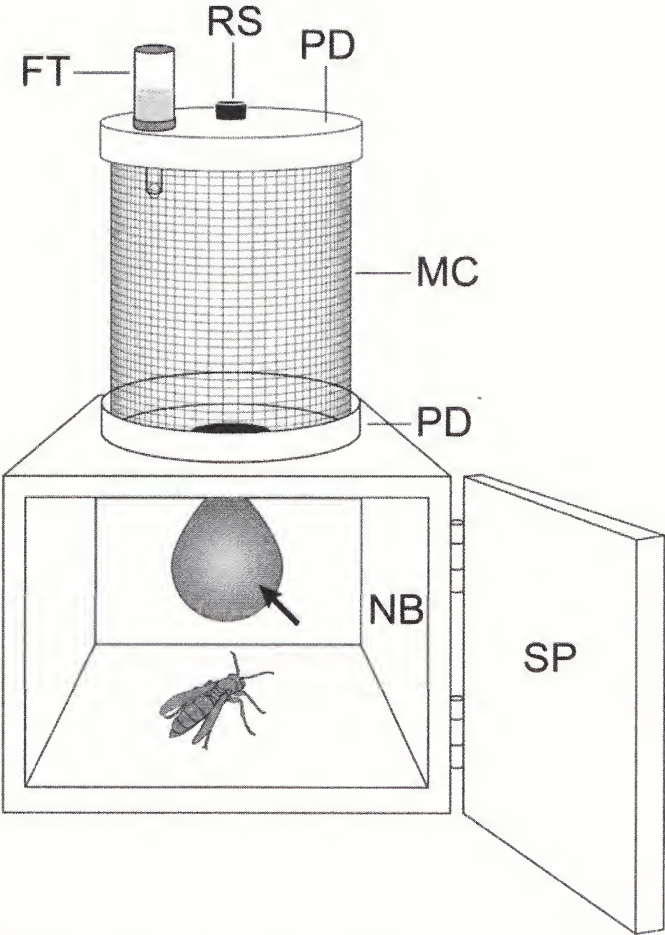


Figure 1. Graphical illustration of a nest box (NB; 15 cm high \times 15 cm wide \times 30 cm long) with hinged side panel (SP) connected to a mesh-cylinder (MC) foraging arena (15 cm in diam \times 20 cm tall), the top and bottom of which reinforced by Petri dishes (PD) for stability and to accommodate a 50-mL falcon tube (FT) with a cotton-filled pipet tip as a water reservoir. The “feeding hole” in the top Petri dish was closed with a rubber stopper (RS) and allowed intermittent insertion of live blow fly and caterpillar prey. The arrow depicts an embryo nest started by the queen.

The effect of collection date on nest initiation could not be tested statistically, because dates could not be assigned to those wasps that failed to initiate a nest. Of the queens we had captured on 15, 17, and 29 April 2014 (57% of the total), six, three, and three, respectively, initiated a nest.

Attachment site of nest pedicel

There was no significant difference in the proportion of queens that attached the nest pedicel to a twig or the roof of the nest box (Figure 2a,b,c; $p=0.77$, FET), between the four species that initiated a nest (Table 2). Of the nest-initiating queens, three of four *V. pensylvanica* queens, three of five *V. alascensis* queens, and five of 11 *V. germanica* queens attached the nest pedicel to a twig. The single nest-initiating *V. acadica* queen did the same.

Within each of the three species (*V. alascensis*, *V. germanica*, *V. pensylvanica*) in which more than one queen initiated a nest, there was no significant deviation from a 50/50 chance in the proportion of queens that attached the nest pedicel to a twig or the roof of the nest box [*V. alascensis*: binomial exact test, $p = 1.0$; *V. germanica*: X^2 goodness-of-fit (1, $N = 11$) = 0.09, $p = 0.76$; *V. pensylvanica*: binomial exact test, $p = 0.63$].

Cells built by queens

Of the five species we studied, queens of four species initiated nests. Of nest-building queens, the mean number of cells they had built (*V. acadica*: 8.0, $N=1$; *V. alascensis*: 17.8 ± 6.3 , $N=5$; *V. germanica*: 21.6 ± 4.6 , $N=11$; *V. pensylvanica*: 26.5 ± 8.3 , $N=4$; Table 1) did not differ at the time we terminated the study [$F_{(3,17)} = 0.502$, $p = 0.686$]. One queen each of *V. alascensis* and *V. germanica* constructed only the nest pedicel and quickly abandoned further attempts of nest building. When we terminated the study (July 20), all larvae and workers had died, and queens had suspended any further nest-building attempts. All queens were dead by mid-July or early August.

Workers produced

Of the 66 queens in our study, each of two *V. germanica* queens maintained a nest that produced worker wasps (two worker wasps from one nest, and six from the other), and one *V. pensylvanica* queen produced a nest from which one worker emerged. All workers appeared at times when the first feral workers appeared in the field, around the first week of June.

DISCUSSION

The differences in nest-initiation rates that we observed between queens of *V. atropilosa* and *V. germanica* (Table 2) reflect the ecological diversity of the genus *Vespula* (Akre *et al.* 1980; MacDonald *et al.* 1980; Akre 1982; Macdonald and Matthews 1984; Landolt *et al.* 1998).

In Pullman (Washington, USA), queens of *V. atropilosa* begin nesting on average 10 days earlier than queens of *V. pensylvanica* (Akre *et al.* 1976). Nests of *V. atropilosa* also decline one month earlier than the nests of most, if not all, members of the *Vespula vulgaris* group (Akre *et al.* 1976). We captured all queens of *V. atropilosa*, which invariably failed to establish nests (Table 2), in late spring (May 15), possibly at a time when these queens could have had established a nest already or could have been tending a nest at an embryo stage, thereby resulting in no (repeated) nest-initiation attempts in our study. However, rearing of *V. atropilosa* nests from over-wintered field-collected queens has never been attempted before, and we may have simply failed to provide one or more essential requisites for successful nesting. Therefore, it remains inconclusive whether *V. atropilosa* queens cannot be reared using the method described here or whether we simply captured *V. atropilosa* queens too late in the season.

We report the first account of nest initiation in a nest box for *V. acadica* and the second account of nest initiation for a member of the *V. rufa* group, the first account being *V. vidua* (Ross *et al.* 1981; Matthews *et al.* 1982; Ross 1983). In our study, the nest initiated by one of four *V. acadica* queens stood out from all other *Vespula* nests in that the queen incorporated prey body

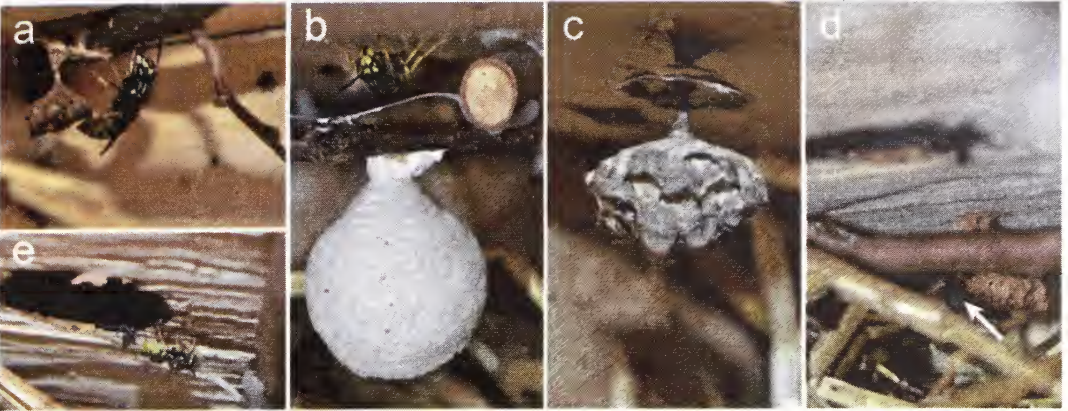


Figure 2. Nest of *Vespula acadica* attached to a twig (surrogate root) in the nest box guarded by the queen (note black pieces of prey incorporated into the nest); (b, c) Embryo nests of *V. germanica* with envelope (b) and of *V. pensylvanica* without envelope (c); (d, e) nest of *V. germanica* with one worker wasp (arrow) tending eggs and larvae (d) and one worker wasp wing fanning at the entrance of the nest box (e).

parts in the nest (Figure 2a). Whether this is typical for *V. acadica* queens will become apparent in further rearing studies or careful inspections of feral nests.

Nest-initiation rates of 61% and 50%, respectively, by queens of *V. germanica* and *V. alascensis* (formerly *V. vulgaris*) in our study (Table 2) were twice as high as those previously reported for these two species (Ross *et al.* 1981) or for *V. germanica* (Leathwick 1997). Conversely, relatively fewer queens of *V. pensylvanica* initiated nests in our study (Table 2) compared to a previous study (Vetter and Visscher 1995). The underlying mechanisms contributing to this differential rearing success are difficult to determine. Unlike previous studies where rearing units resided indoors with small temperature oscillations and a constant photoperiod (Ross *et al.* 1981; Vetter and Visscher 1995; Leathwick 1997), we kept our nest boxes outdoors and thus exposed them to seasonal changes in photoperiod and to significant diel and seasonal temperature fluctuations. However, the straw inside the nest boxes that we provided as insulation material may have been insufficient to keep *V. pensylvanica* queens warm and to induce more consistent nest building. The relatively high propensity of *V. germanica* queens to initiate nests irrespective of rearing conditions might be an intrinsic characteristic of *V. germanica* that may help explain why this wasp is so widely distributed and invasive in North and South America as well as in New Zealand (MacDonald *et al.* 1980; D'Adamo *et al.* 2001; Brown *et al.* 2014). Both feral and laboratory-reared nests of *V. germanica* have the fastest nest-development rates of all species studied in North America (MacDonald *et al.* 1980; Matthews *et al.* 1982).

The type of potential attachment sites for nest pedicels does not seem to matter critically, because the same number of queens attached the nest pedicel to the roof of the nest box or to a twig serving as surrogate root in a quasi-subterranean nest cavity. Considering, however, that the surface of roots was much smaller than the surface of nest box roofs, queens may indeed have preferred roots as potential attachment site for nest pedicels. Alternatively, the preference for pedicel attachment sites may vary between queens. If so, providing diverse and multiple sites for pedicel attachment could help increase rates of nest initiation.

The rate of cell building reflects queen quality and varies with species (Archer 2009). In our study, we could not consistently track nest development such as cells built per day, eggs laid, and number of cells with larvae or pupae, because 35% of the queens that initiated a nest built an envelope surrounding the cells (Figure 2c). Vetter and Visscher (1995) faced the same challenge with one of the four *V. pensylvanica* nests they reared. At the end of our study, however, we did record the number of cells per nest and did not find a significant difference in the mean number of cells built between species (Table 2). Apparently, all but three nesting activities (see below) were discontinued at the same point of brood development, just before the emergence of the first

worker wasps that would have continued all tasks except egg laying (Gambino and Loope 1992). Eggs and larvae died from unknown causes. How and why two *V. germanica* and one *V. pensylvanica* queens progressed to producing a few worker wasps (Figures 2d,e) remains unknown. We envision that the well-being of larvae could have been compromised by a fungal pathogen, although fungal growth was not apparent on food remains such as legs and wings of fed-on house flies that we left in the feeding cage. Alternatively, larvae may have suffered from a lack of nutritional diversity or key nutrients. Conceivably, free foraging eusocial wasps self-medicate in that they adjust their diet, or that of their offspring, in response to pathogens, as do caterpillars of *T. ni* and *Grammia incurrupta* (Edwards) (Singer *et al.* 2009; Shikano and Cory 2014).

CONCLUSION

Queens of the five *Vespula* species that we attempted to rear in nest boxes differed in nest-initiation rates, with *V. germanica* having a greater success rate than *V. atropilosa*. Whether these differences are due to intrinsic characteristics of these species, external factors such as ambient temperature during rearing, or the quality of the queens we had collected in the spring cannot be ascertained. The high propensity of *V. germanica* queens to initiate nests may be a contributing factor to the success of *V. germanica* as one of most pestiferous and invasive wasp species worldwide.

Most nests in our study failed to produce worker wasps. We speculate that these nests succumbed to a pathogen rather than to faulty rearing methodology, because all larvae visible in those nests that ceased to develop started to die within days of each other and showed similar signs of a fungal infection. We recommend that, in future attempts to rear yellowjackets, queens are allowed to forage freely as soon as they have initiated nest building. This would enhance the nutritional diversity for larval offspring, provide the essential nutrients at particular times during nest development, and possibly help curtail the effect of pathogens in the food or nest. Regardless, perfecting techniques for rearing yellowjackets in further studies is well justified, because it will greatly increase the opportunity of investigating the role of these intriguing predatory insects in ecosystems and the evolution of eusociality in the Hymenoptera.

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Habitat associations of adult Oregon branded skipper, *Hesperia colorado oregonia* (W. H. Edwards, 1883) (Lepidoptera: Hesperiiidae), at Cordova Shore, Vancouver Island, British Columbia

L. L. GELLING¹

ABSTRACT

Oregon branded skipper, *Hesperia colorado oregonia* (W. H. Edwards, 1883), is a nationally (Canada) endangered butterfly extant at six remaining sites on Vancouver Island, British Columbia, Canada. I studied the habitat associations of this subspecies within a coastal sand ecosystem at Cordova Shore, on southeast Vancouver Island. Little is known about habitat requirements for this butterfly; however, because of its endangered status, information on its habitat is required to help direct conservation action. During the summers of 2013 and 2014, most of the butterflies observed (91%) were found within the dune wildrye-beach pea (*Leymus mollis*–*Lathyrus japonicus*) terrestrial ecosystem, with 9% observed in the black knotweed–yellow sand verbena (*Polygonum paronychia*–*Abronia latifolia*) terrestrial ecosystem. To determine the habitat characteristics preferentially selected by the adult butterflies, I compared occupied and simultaneously unoccupied sites. I used conditional logistic regression for matched pairs to investigate relationships between butterfly presence and six habitat variables in 21 occupied and 21 random plots, and used Akaike information criterion (AIC) to identify the best among a set of 20 candidate models. The best-supported model included black knotweed (*Polygonum paronychia*) and Oregon gumweed (*Grindelia oregonia*) as the sole variables predicting the occurrence of Oregon branded skipper. The model predicted that the likelihood that Oregon branded skipper would be present increases with increased cover of Oregon gumweed (OR = 1.5, 95% CI: 0.9–2.6) and black knotweed (OR = 1.6, 95% CI: 0.1–23.6). The two plants are likely important for the skipper as adult nectar sources; however, small sample sizes and model confidence intervals suggest caution should be used when applying the model. In light of these findings, I provide guidance for future conservation of Oregon branded skipper to land managers within Cordova Shore.

Key Words: Lepidoptera, *Hesperia*, *Hesperia colorado oregonia*, habitat, endangered, British Columbia

INTRODUCTION

About one-third of butterfly species in Canada are at some level of risk from threats such as habitat loss and degradation, pesticides, invasive species, and habitat changes resulting from climate change (Hall 2009). To effectively direct conservation actions towards at-risk butterflies, an understanding of their basic biological needs is required. For many butterflies, basic biology (including resource needs) is not well known (Schultz and Crone 2008). Butterflies have four life stages (egg, caterpillar, pupa, adult), and the resources required to complete each life stage can differ. I examined two aspects of the biology of the adults of a rare butterfly, Oregon branded skipper, *Hesperia colorado oregonia* (W. H. Edwards); ecosystem selection and habitat components used within the ecosystem.

In 2013, the Committee on the Status of Endangered Wildlife in Canada (COSEWIC) designated Oregon branded skipper as endangered based on historic loss of habitat and other threats (COSEWIC 2013). Habitat of the skipper includes Garry oak (*Quercus garryana* Douglas ex Hook) associated ecosystems and sparsely vegetated coastal sand ecosystems (COSEWIC 2013). These ecosystems currently contain many rare and at-risk plant species and communities (BCCDC 2014). Presently, the greatest threats to this subspecies include vegetation succession of

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open habitats and the potential spraying of *Bacillus thuringiensis* var *kurstaki* (Btk) pesticide to control *Lymantria dispar* Linnaeus, the invasive gypsy moth (COSEWIC 2013). The subspecies is considered at risk globally (G5T3T4; NatureServe 2008) and is on the British Columbia red list (S1; BCCDC 2014).

Oregon branded skipper occurs from California north through west-central Oregon, into the Puget Trough of the state of Washington and onto the southern tip of Vancouver Island in southeastern British Columbia, Canada. The subspecies is known from 20 sites on Vancouver Island, from Greater Victoria north to Cameron Lake. As of 2014, populations remain extant at only six of these sites (Figure 1; Table 1).

The natural history and habitat associations of Oregon branded skipper are not well studied. Related skippers are known to inhabit areas with exposed bare ground and dry, well-drained soil patches (e.g., *Hesperia comma*, Thomas *et al.* 1986; *Hesperia assiniboia*, COSEWIC 2013, and; *Polites mardon*, Pyle 2002). Branded skipper (*Hesperia* spp.) larval foodplants are typically monocotyledons, grasses and sedges (Layberry *et al.* 1998), but foodplants specific to the Oregon branded skipper subspecies are not known. In one case on Vancouver Island, reared larvae consumed hand-fed grasses from the genera *Lolium* and *Bromus* (Hardy 1954). Oviposition substrate is not necessarily good evidence of larval foodplants, because *Hesperia* will oviposit on other substrates near the host, including fence posts and tree trunks (MacNeill 1964; Pyle 2002). The flight period of Oregon branded skipper is early July to mid-September (Layberry *et al.* 1998; Guppy and Shepard 2001). The species has one generation per year. Oviposition occurs in the summer; eggs overwinter from September to spring and hatch between March and April. Larvae emerge and feed from spring to summer, developing through six instar stages before the pupal (chrysalis) stage, which occurs from early July to late August (Hardy 1954; James and Nunnallee 2011).

To better understand habitat associations of the adult stage of Oregon branded skipper, I investigated relationships between (1) butterfly presence and ecosystem type, and (2) butterfly presence and vegetation and substrate cover. The study area was at “Cordova Shore”, the only coastal sand ecosystem in British Columbia from which Oregon branded skipper is known. Cordova Shore is a 437-ha area on southeast Vancouver Island, and contains remnants of rare and

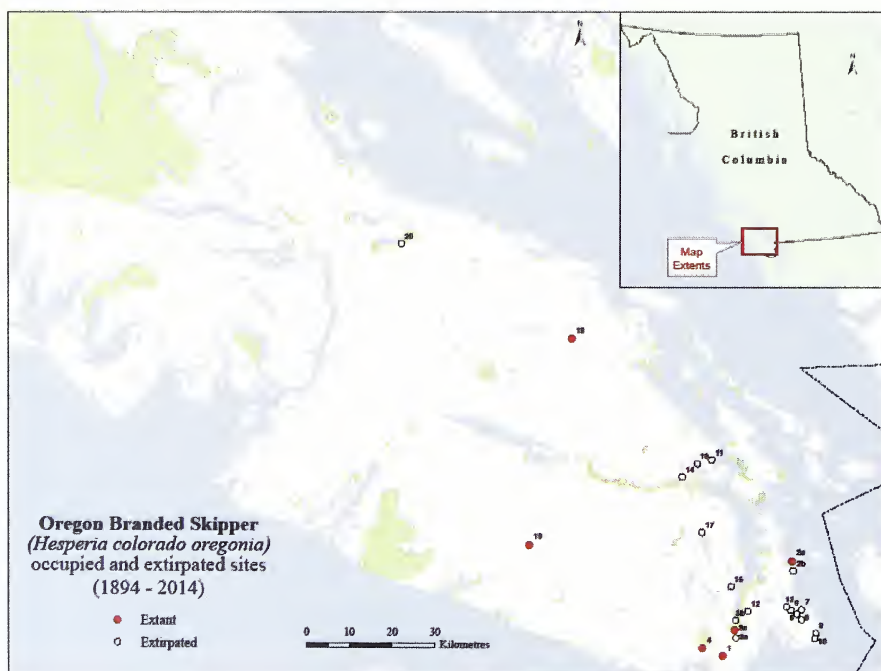


Figure 1. Oregon branded skipper-occupied and -extirpated sites (1894–2014).

Table 1

Historical and current (2014) locations of Oregon branded skipper. (RBCM collection = Royal British Columbia Museum; CDC (2014) = record housed within the BC Conservation Data Centre (<http://www.env.gov.bc.ca/atrisk/toolintro.html>); * = the UTM coordinate of the historical record has been estimated for mapping purposes.) The data from this table has been archived: (<http://dx.doi.org/10.6084/m9.figshare.1569875>)

Site	UTM Zone	Easting	Northing	Site Name	Year Last Observed	Current Viability	Source of Record
1	10	455737	5360927	Camas Hill, Metchosin	2011	Extant (COSEWIC 2013)	COSEWIC 2013
2a	10	472597	5382583	Cordova Spit; Saanich	2014	Extant (COSEWIC 2013)	CDC (2014)
2b	10	472927	5380216	Island View Beach Regional Park; Saanich*	1963	Extirpated (COSEWIC 2013)	RBCM collection
3a	10	459000	5365000	Goldstream; Mount Wells Regional Park*	1953	Extirpated (COSEWIC 2013)	RBCM collection
3b	10	459095	5369096	Goldstream Provincial Park*	1952	Extirpated (COSEWIC 2013)	RBCM collection
3c	10	458694	5367074	Goldstream; Humpback Road area	2009	Extant (COSEWIC 2013)	COSEWIC 2013
4	10	451123	5362965	Mt. Manuel Quimper Capital Regional District Park	2014	Extant (COSEWIC 2013)	CDC (2014)
5	10	473400	5370000	Blenkinsop Lake*	1951	Extirpated (COSEWIC 2013)	RBCM collection
6	10	472000	5371000	Rithets Bog; Saanich Park*	1956	Extirpated (COSEWIC 2013)	RBCM collection
7	10	474570	5371016	Mount Douglas Saanich Park*	1953	Extirpated (COSEWIC 2013)	COSEWIC 2013
8	10	474400	5368600	Braefoot; Saanich*	1953	Extirpated (COSEWIC 2013)	RBCM collection
9	10	477600	5365300	Oak Bay (Uplands Park)*	1953	Extirpated (COSEWIC 2013)	RBCM collection
10	10	477324	5364029	Oak Bay (Victoria area)*	1953	Extirpated (COSEWIC 2013)	RBCM collection
11	10	455000	5407000	Maple Bay*	1935	Extirpated (COSEWIC 2013)	RBCM collection
12	10	462000	5371200	Langford, Millstream Road*	1955	Extirpated (COSEWIC 2013)	RBCM collection
13	10	471100	5371800	Royal Oak; Observatory Hill; Saanich*	1955	Extirpated (COSEWIC 2013)	RBCM collection
14	10	447992	5403280	Duncan*	1926	Extirpated (COSEWIC 2013)	RBCM collection
15	10	458332	5377088	Malahat*	1920	Extirpated (COSEWIC 2013)	RBCM collection
16	10	451675	5406270	Quamichan Lake*	1917	Extirpated (COSEWIC 2013)	COSEWIC 2013
17	10	452076	5390053	Shawnigan Lake*	1894	Extirpated (COSEWIC 2013)	RBCM collection
18	10	423541	5436827	Nanaimo River Road	2011	Extant	C. Guppy, personal collection
19	10	411728	5388924	Port Renfrew	2014	Extant	Photo by M. Yip and confirmed by C. Guppy
20	10	5460600	384800	Cameron Lake*	1952	Undetermined	RBCM collection

sensitive sand dune and estuarine ecosystems (Stacey and Filatow 2009). It has been—and continues to be—affected by changes to natural ecological processes and vegetation by hydrological forces, recreation, invasive species, disruption to coastal sediment transport processes, and development (Page 2010). Oregon branded skipper has been reported from Cordova Shore since the 1950s, from two locations labelled “Cordova Spit” (called TIXEN by Tsawout First Nation) and “Island View Beach” (Table 1). Inventory from 2001 to 2014 confirmed the skipper’s continued existence at Cordova Spit, but the species has not been reported from Island View Beach since 1963. A better understanding of the habitat relationships of this species will aid in management and conservation for this endangered butterfly within Cordova Shore.

MATERIALS AND METHODS

The study site was a 36-ha area within Cordova Shore on the east side of the Saanich Peninsula on Haro Strait (Figure 2a). At the north end is a sandspit, which is bordered on the west by Saanichton Bay and on the east by Cordova Channel. South of the spit is a coastal inland area surrounded by residential housing and agricultural fields. The site consists of a variety of broad ecosystem types, including coastal sand dune, beach, wetland, estuary and forest, with many smaller ecosystems contained within them. It is under the jurisdiction of three landowners: Tsawout First Nation, the District Municipality of Central Saanich, and the Capital Regional District (Island View Beach Regional Park; Figure 2b).

Butterflies are most active when they have warmed from the sun (Guppy and Shepard 2001); thus, I surveyed between approximately 10:00 and 16:00 hours under conditions ranging from full sun to overcast days with air temperatures above 19°C. To locate Oregon branded skipper, one or two surveyors searched for butterflies by walking along a systematic, continuous transect that consisted of a series of parallel east–west oriented survey lines 25 m apart. The survey transect extended from the north tip of TIXEN/Cordova Spit to the south end of the Island View Beach parking lot. The transect was surveyed in alternate directions; from north to south on one survey, and from south to north on the subsequent survey. Because one full transect took more than one day to complete, I used a GPS coordinate to mark my finish location at the end of each day and resumed the route on the next field day.

When an Oregon branded skipper was observed, I captured it with a net to confirm identification, and then released it. The very similar woodland skipper, *Ochlodes sylvanoides* (Boisduval, 1852), was often abundant and made identification of Oregon branded skipper difficult until specimen capture. I recorded the plant or substrate it was using or landed on (if initially observed in flight) and established a 5m² plot centred at each observation point. I recorded and measured the percent cover of the abiotic and biotic variables within each plot (Table 2). For each plot in which a butterfly was recorded, I established a second, paired plot, located in a random compass direction and distance (up to 30 m from the butterfly detection point). This plot was sampled using the same methods. I overlaid the geo-coordinates of all butterfly locations on existing terrestrial ecosystem maps (Stacey and Filatow 2009) to determine which ecosystem units they occurred in.

I evaluated habitat factors that affected the probability of a site being used by an Oregon branded skipper using a variety of inferential techniques. I used an information-theoretic approach to identify the model from a candidate set that best predicted the occurrence of butterflies at a site, based upon a variety of habitat factors. To select variables for model building, I calculated correlation coefficients among all measured habitat variables and screened out variables that were too highly correlated to retain in the same model (coefficients of <0.4 or >0.4; Ballinger 2004). I also excluded variables that did not show a significant difference in percent cover between occupied and random plots (i.e., $P \geq 0.05$). I retained variables that were previously thought to be important for *Hesperia* spp. (monocotyledons, grasses and bare ground) and those I observed being used by the butterfly during field observation. The final variables selected for the analysis were 1) Oregon gumweed (*Grindelia stricta*), 2) black knotweed (*Polygonum paronychia*), 3) yarrow (*Achillea millefolium*), 4) red fescue (*Festuca rubra*), 5) moss spp., and 6) sand.

Table 2

Percent cover (standard deviation) and origin for all variables measured in occupied and random plots. Variables with * were retained for statistical analysis.

Plant Species/ Substrate	Origin	Occupied	Random	Plant Species/ Substrate	Origin	Occupied	Random
Abronia latifolia	native	1(4)	<1 (0.2)	Leymus mollis ssp. mollis	native	20(17)	20(28)
Achillea millefolium var pacifica	native	2(2)	2(4)	Lomatium nudicaule	native	<1(1)	<1(0.1)
Aira caryophyllea	non-native	<1(0.7)	0	Malus fusca	native	1(3)	1(5)
Aira praecox	non-native	2(6)	<1(1.5)	moss spp. *	undetermined	26(17)	35(20)
Ambrosia chamissonis *	native	5(10)	3(8)	Plantago maritima ssp. juncoides	native	0	<1(0.2)
Ammophila arenaria	non-native	0	<1(0.2)	Poa pratensis	non-native		<1(0.2)
Anthoxanthum odoratum	non-native	<1(0.9)	<1(0.1)	Polygonum paronychia *	native	8(13)	2(5)
Armeria merittima var maritima	non-native	3(9)	<1(1)	Puccinellia nutkaensis	native	0	3(9)
Atriplex gmelinii	native	<1(0.2)	<1(0.5)	Rosa rugosa	non-native	<1(2)	<(0.2)
Cakile edentula	non-native	0	<1(0.7)	Rubus armeniacus	non-native	2(10)	2(7)
Calystegia soldanella	native	<1(2)	<1(0.1)	Rumex acetosella	non-native	1(7)	<1(1.1)
Carex macrocephala	native	3(9)	8(19)	Sarcocornia pacifica	native	<1(1.5)	4(13)
Cytisus scoparius	non-native	1(2)	2(3)	Vicia nigricans ssp. gigantea	native	0	<1(0.1)
Dactylis glomerata	non-native	<1(0.1)	0	Vulpia microstachys var. pauciflora	native	0	<1(0.7)
Distichlis spicata var spicata	native	4(10)	1(5)	sand *		17(14)	25(34)
Festuca rubra ssp. rubra *	non-native	19(21)	12(20)	pebbles		<1(0.2)	<1(0.5)
Grindelia stricta *	native	9(8)	2(4)	cobble		<1(0.1)	<1(2.2)
Holcus lanatus	non-native	1(1)	2(6)	coarse woody debris		2(4)	8(10)
Honckenya peploides ssp. major	native	0	<1(0.1)	garbage		4(13)	0(0)
Hypochaeris glabra	non-native	0	<1(0.1)	short turf grass		3(6)	<1(1.5)
Hypochaeris radicata	non-native	<1(0.9)	<1(0.1)	long grass		41(26)	36(35)
Lathyrus japonicus var maritimus	native	0	<1(0.2)	shrubs		12(2)	59(8)
Lepidium densiflorum	native	<1(0.3)	<1(0.1)				

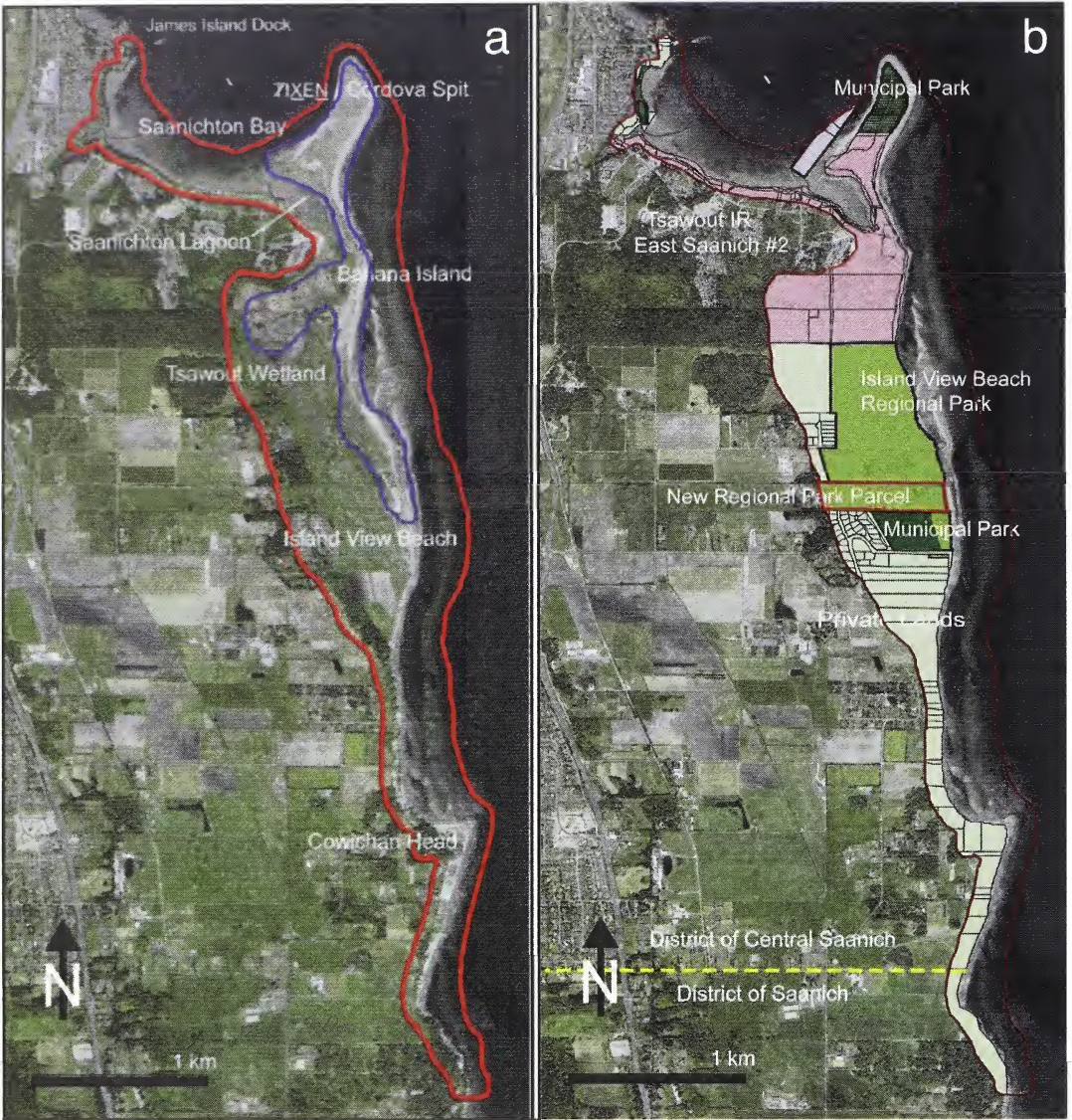


Figure 2a. Cordova Shore boundary. East side of the Saanich Peninsula on Haro Strait, Vancouver Island, British Columbia (exterior outline). The interior outline is the study area. Maps from Cordova Shore Conservation Strategy (with permission).

Figure 2b. Land ownership and property boundaries within Cordova Shore: Tsawout First Nation, CRD Regional Parks (Island View Regional Park), Central Saanich municipal parks, and private property.

I used conditional logistic regression for matched pairs to investigate the relationships between the six selected variables in both occupied and random plots. I utilized combinations of up to two variables (one variable for each of the 10 observations) as per Peduzzi *et al.* (1996), resulting in 20 hypothetical models that I tested with information-theoretic methods using Akaike information criterion (AIC; Table 3). This method does not assume that the true model is in the set; however, it provides the best fit candidate model, based on thoughtful selection of variables (Burnham and Anderson 1998). I conducted all statistical analyses in R (R Core Team 2014).

Table 3

Candidate models evaluated for predicting Oregon branded skipper presence at Cordova Shore. Model scores are ranked based on relative Akaike’s Information Criterion values (Delta_AIC). The number of parameters (K), log likelihood value (LL), AIC-corrected for small-sample bias (AICc); likelihood function of the model (ModelLik); AIC weights (AICcWt), and; cumulative weights (Cum.Wt) are reported for each model.

Model	K	AICc	Delta_AICc	ModelLik	AICcWt	LL	Cum.Wt
<i>Polygonum paronychia</i> + <i>Grindelia oregonia</i>	2	8.943	0.000	1.000	0.928	−2.284	0.928
<i>Polygonum paronychia</i> + <i>Ambrosia chamissonis</i>	2	16.261	7.318	0.026	0.024	−5.943	0.952
<i>Festuca rubra</i> + <i>Grindelia oregonia</i>	2	18.020	9.077	0.011	0.010	−6.823	0.961
moss_cover + <i>Grindelia oregonia</i>	2	18.175	9.231	0.010	0.009	−6.906	0.971
<i>Polygonum paronychia</i>	1	18.559	9.616	0.008	0.008	−8.219	0.978
<i>Festuca rubra</i> + <i>Polygonum paronychia</i>	2	20.159	11.216	0.004	0.003	−7.892	0.982
<i>Festuca rubra</i> + <i>Ambrosia chamissonis</i>	2	20.185	11.242	0.004	0.003	−7.905	0.985
sand + <i>Polygonum paronychia</i>	2	20.378	11.435	0.003	0.003	−8.001	0.988
<i>Grindelia oregonia</i>	1	20.558	11.615	0.003	0.003	−9.220	0.991
moss_cover + <i>Polygonum paronychia</i>	2	20.610	11.667	0.003	0.003	−8.118	0.994
sand + <i>Grindelia oregonia</i>	2	20.910	11.967	0.003	0.002	−8.273	0.996
<i>Festuca rubra</i>	1	22.017	13.074	0.001	0.001	−9.948	0.997
<i>Grindelia oregonia</i> + <i>Ambrosia chamissonis</i>	2	22.757	13.814	0.001	0.001	−9.197	0.998
<i>Festuca rubra</i> + moss_cover	2	23.582	14.639	0.001	0.001	−9.603	0.999
sand + <i>Festuca rubra</i>	2	24.162	15.218	0.000	0.000	−9.893	0.999
moss_cover	1	25.671	16.728	0.000	0.000	−11.777	0.999
sand	1	25.833	16.889	0.000	0.000	−11.858	1.000
<i>Ambrosia chamissonis</i>	1	26.335	17.391	0.000	0.000	−12.108	1.000
moss_cover + <i>Ambrosia chamissonis</i>	2	27.232	18.289	0.000	0.000	−11.434	1.000
sand + <i>Ambrosia chamissonis</i>	2	27.312	18.368	0.000	0.000	−11.474	1.000

RESULTS

I conducted surveys on 24 days between 16 July and 13 September 2013. In 2014, I revisited the Cordova Spit on three days to ensure that the population remained extant and to collect additional data (30 July and 4, 5 September 2014). I recorded 22 Oregon branded skipper in 21 separate plots (20 in 2013, and 2 in 2014). Based on terrestrial ecosystem map boundaries and field verification, most occurrences (91%) were within the dune wildrye–beach pea (*Leymus mollis*–*Lathyrus japonicus*) ecosystem unit (Stacey and Filatow 2009). Two skippers (9%) were

Table 4

Odds ratio coefficients (OR), standard errors (SE), and 95% confidence intervals for black knotweed and Oregon gumweed variables used in the final model.

Variable	OR	SE	C.I.
Black knotweed	1.6	1.38	0.1–23.6
Oregon gumweed	1.5	0.28	0.9–2.6

detected in the black knotweed–yellow sand verbena (*Polygonum paronychia*–*Abronia latifolia*) ecosystem to the north (Figure 3). I observed skippers primarily between 12:00 and 16:00. One individual was seen at 09:20. I observed them nectaring and resting primarily on Oregon gumweed, but they also rested on yarrow (*Aquillea millefolium*), moss spp, black knotweed, red fescue, seashore saltgrass (*Distichlis spicata*), and sand.

The probability of use by Oregon branded skipper was related to the cover of several habitat features. The best model from the candidate set was that which predicted probability of use from the cover of black knotweed and Oregon gumweed, which scored 8 AIC units better than the next best model (Table 3). The relationship was positive (both odds ratio coefficients were greater than 1); however, because both 95% confidence intervals included 1 and the standard error of the black knotweed odds ratio was high, the relationship was not conclusive. The best model estimated that the probability of use by Oregon Banded Skipper increased with increasing cover of both black knotweed (OR: 1.6, 95% CI: 0.1–23.6) and Oregon gumweed (OR: 1.5, 95% CI: 0.9–2.6; Table 4). An odds ratio of 1.6 for black knotweed suggested that a 1% increase in cover increased the odds of use by a butterfly by 60% (1.6 times higher than the odds of a butterfly not being present). For Oregon gumweed, an odds ratio of 1.5 suggested that a 1% increase in cover increased the probability of use by a butterfly by 50% (1.5 times higher than the odds of a butterfly not being present). Occupied plots also had a higher mean percent cover of black knotweed than in random

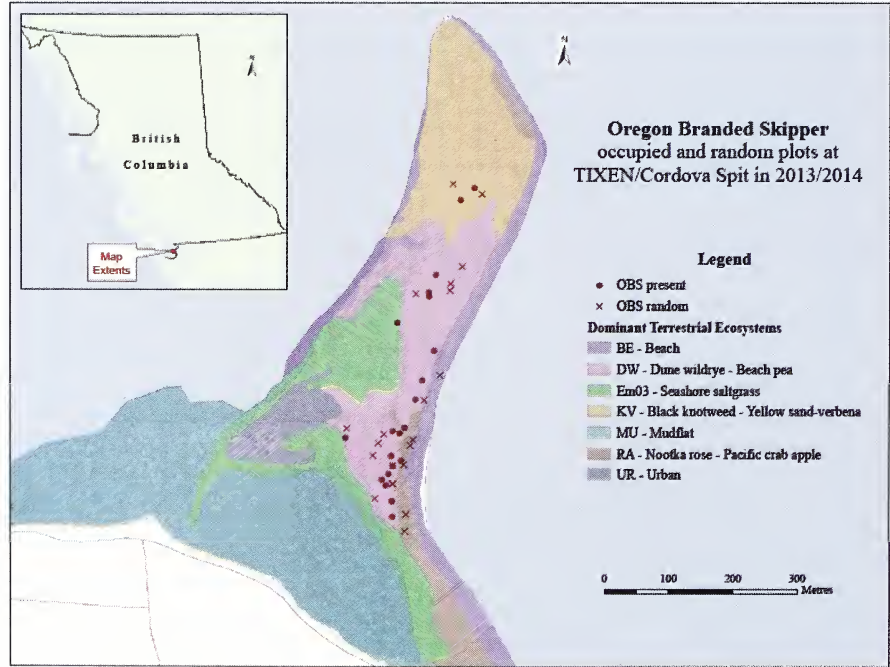


Figure 3. Oregon branded skipper-occupied and random plots at TIXEN/Cordova Spit in 2013/2014. Ecosystem spatial data from Stacey and Filatow (2009).

plots (paired t-test: 8%; $t = 2.12$; $df = 20$; $p = 0.04$). Occupied plots had a higher mean percent cover of Oregon gumweed than random plots (paired t-test: 9%; $t = 3.32$; $df = 20$; $p = 0.003$).

DISCUSSION

My results suggest that Oregon branded skipper is more likely to occupy ecosystems at TIXEN/Cordova Spit where black knotweed and Oregon gumweed occur together. Oregon gumweed is a common perennial herbaceous species found on beaches, rocky bluffs, and disturbed sites (BCCDC 2014; E-Flora 2014). This plant flowers throughout the summer and likely plays an important role in providing a nectar source for Oregon branded skipper and other butterflies within Cordova Shore. Black knotweed is a small, low-lying, flowering perennial shrub that is restricted to moist sand dunes and beaches on southern Vancouver Island and the Gulf Islands (BCCDC 2014; E-Flora 2014). Black knotweed also likely provides nectar for Oregon branded skipper.

Factors other than black knotweed and Oregon gumweed may affect the probability of use by the butterfly. I found only two individuals within the black knotweed–yellow sand verbena ecosystem that makes up a large part of the north end of TIXEN/Cordova Spit (Figure 3). Although this area contains both black knotweed and Oregon gumweed, it is exposed to the elements, with bare, shifting sand, erosion and deposition from wind and waves (Stacey and Filatow 2009). These conditions may be favorable for the plants; however, they may be too volatile for the butterfly. Most skippers were observed within the dune wildrye–beach pea ecosystem in the central part of the spit. Within this large ecosystem, a smaller unit of black-knotweed–yellow sand verbena contains Oregon gumweed and black knotweed. The butterflies appear to be concentrated there. To the east of this unit, paralleling the beach, there is a natural slope with dense tall grasses, blackberry, rose, and crabapple. This may have created a favorable



Figure 4. Locations of black knotweed at Cordova Shore. Map, with permission, from the Cordova Shore Conservation Strategy (Page 2010). Photo is south of the boundary between Tsawout First Nations and Capital Regional District lands.

patch of habitat for the butterflies, in that they can utilize the plants while remaining protected by the slope from wind and sand deposition coming off the ocean.

Bunchgrasses, such as red fescue (*Festuca rubra*), are suspected to be larval foodplants of Oregon branded skipper (COSEWIC 2013). Red fescue is a characteristic component of the dune wildrye–beach pea ecosystem (Stacey and Filatow 2009), in which the majority of skippers occurred. Based on the model analysis, red fescue did not appear to have a significant effect on the probability of the skipper being present (Table 3). This may be because adults tend to focus on nectar sources, and only females actively ovipositing would potentially use red fescue. No ovipositing females, eggs, or larvae were observed during this study.

Cordova Spit is one of best intact examples of the extremely rare coastal dune ecosystems on Vancouver Island (Page 2010). The Oregon branded skipper population at the spit has persisted since at least 1952. Hill *et al.* (1996) investigated *Hesperia comma* metapopulations in 69 patches over 9 years in Surrey, United Kingdom. They found that local populations in small (<1 ha), isolated patches were more likely to go extinct than if the patches were large and close (<1 km) to other patches. The patch at TIXEN/Cordova Spit is 6 ha. This is apparently large enough to sustain a population of Oregon branded skipper even without another nearby population. Oregon branded skipper is not migratory, and maximum dispersal distance is unknown. Rescue from other populations is unlikely as the closest extant population is the Goldstream site, 18 km southwest of TIXEN/Cordova Spit. No other known extant populations in British Columbia occur in coastal sand ecosystems. In the U.S.A., the closest population is 40 km east of Cordova Spit, at Orcas Island, Washington (A. Potter, pers. comm. 2013). Populations on San Juan Island (Washington, U.S.A.) are now extirpated (COSEWIC 2013).

MANAGEMENT RECOMMENDATIONS

Cordova Shore is the only known coastal sand ecosystem in British Columbia that supports Oregon branded skipper. Future monitoring on Cordova Spit should focus on the ecosystems in which the species was recorded in this study. To retain and recover this species, it is likely necessary to protect the remaining habitat and restore what has been degraded. Implementing the recommendations from the Cordova Shore Conservation Strategy to maintain sparsely vegetated sandy habitats by removing invasive plants, sustaining sand movement, and reducing intensive recreation (Page 2010) should aid efforts to conserve and recover this butterfly subspecies. Property managers should focus on restoration and protection of ecosystems that contain Oregon gumweed and black knotweed, as well as the likely larval foodplant(s) of red fescue and other native grasses. This can be done by preventing and reducing ecosystem damage, targeting areas for restoration, and managing pesticide application if a gypsy moth outbreak occurs in the area.

Signage can aid in preventing ecosystem damage. Two Tsawout First Nation signs at the parking lot entrance to Cordova Spit likely help protect the butterflies' habitat. One prohibits dogs, horses, camping, fires and dumping and the other brings awareness to sensitive habitat(s) in the area. There is a black knotweed–yellow sand verbena ecosystem containing both Oregon gumweed and black knotweed that straddles the Tsawout First Nation- and CRD-managed lands (Island View Beach; Figure 4). It is partially fenced on the Island View Beach property. There is a single sign at the upper west end of the fence that states that the area is closed for restoration. An additional sign on the fence facing the main path closest to the beach would be beneficial, because it sees the most foot traffic. A Tsawout First Nation sensitive-ecosystem sign at the property line would also improve awareness.

There is fencing near sensitive vegetation and gated barriers to restrict access by off-road vehicles to Cordova Spit, which has allowed natural vegetation to re-establish. On the Island View Beach land, the patch of black knotweed–yellow sand verbena ecosystem is not completely contained by fencing and is surrounded by metre-wide paths (Figure 4). Although the patch is small (~770 m²), it may be the only site at Island View Beach that could potentially provide suitable habitat. Across from this patch, a natural slope with tall vegetation borders the beach, similar to the habitat where the butterflies are currently found. Extending or moving the fence to

include the entire ecosystem, and reducing the number of paths surrounding it, would be helpful in diverting traffic away from this area.

Throughout Cordova Shore, non-native plant species such as European beachgrass (*Ammophila arenaria*), gorse (*Ulex europaeus*), Scotch broom (*Cytisus scoparius*), and Himalayan blackberry (*Rubus armeniacus*) have become established and need to be monitored and controlled to prevent encroachment into the butterflies' habitat.

One of the greatest potential threats to Oregon branded skipper is pesticide application (COSEWIC 2013). *Bacillus thuringiensis* var *kurstaki* (Btk) pesticide, used to control the invasive gypsy moth, has been shown to eliminate the larval stage of non-targeted lepidoptera on Vancouver Island (Guppy and Shepard 2001; Boulton 2004). If a gypsy moth (or other lepidopteran pest) outbreak occurs, it will be important to avoid spraying skipper habitat to avoid harming the larvae.

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Control of Apple Clearwing Moth, *Synanthedon myopaeformis*, with Tree-trunk Applications of Reduced-risk Insecticides, Nematodes and Barriers

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ABSTRACT

Apple clearwing moth (ACM), *Synanthedon myopaeformis* (Borkhausen) (Lepidoptera: Sesiidae), was discovered in an apple orchard in Cawston, British Columbia, in 2005. This xylophagous, European species has become a serious pest in high-density apple orchards where size-controlling rootstocks are attacked by the wood-boring larvae. Seven screening trials with reduced-risk insecticides, including seven conventional treatments and three organic treatments, were established in two commercial, high-density, mixed-variety apple plantings in Cawston. Objectives were: (1) to screen several new synthetic insecticides having novel chemistries that purportedly have reduced risks; (2) to evaluate use of several organically approved spray materials, including nematodes; and, (3) to compare the efficacy of various products when applied at different times during the growing season. Single, handgun sprays delivering treatments in 2000 L of water ha⁻¹ at 200 psi were applied as curative sprays targeting mature larvae in rootstock–scion graft unions in May and October 2008, and in June 2009. Among seven treatments tested, only the insect growth regulator, Rimon® 10 EC (10% novaluron), at 2.8 L of product ha⁻¹, consistently reduced adult emergence compared with untreated control trees in all experiments. When applied twice as *preventative* treatments during flight of male ACM in 2008, Altacor®, Belt®, Delegate™, Entrust® and Rimon all caused significant reductions in adult emergence the following year; the Rimon treatment exhibited the greatest reduction (–96.4%). In a similar 2009 trial, only Rimon reduced populations the following year. One curative application of the organic materials, Entrust®, Crocker’s Fish Oil®, or Purespray Green Oil™, at any spray timing, did not control ACM. Applying *Steinernema feltiae* (Filipjev) at 1×10⁵ infective juvenile nematodes / 100 ml of water / tree provided significant control of ACM in one spring 2008 trial. In two 2009 nematode-only experiments, a sawdust paste tree-trunk barrier applied over nematode treatments made either in May or August caused significant reductions in emergence of ACM adults. Curative tree-trunk sprays of Rimon 10 EC at the tested rate are recommended for control of ACM in conventional apple orchards. Tree-trunk barriers and nematodes warrant further study as possible organic controls for ACM.

Key Words: Lepidoptera, Sesiidae, clearwing borer, invasive species, apples, Rimon, novaluron, curative sprays, preventative sprays, tree-trunk barriers,

INTRODUCTION

Synanthedon myopaeformis (Borkhausen) (Lepidoptera: Sesiidae), the apple clearwing moth (Alford 2007), sometimes called small red-belted clearwing borer (Ateyyat and Al-Antary 2006), is a xylophagous, European insect that commonly infests species of *Rosacea* (Špatenka *et al.* 1999). The first North American detection of *S. myopaeformis* occurred in 2005 (Philip 2006), when a moth was collected in an organic apple orchard in the Similkameen Valley, near Cawston, British Columbia (B.C.). In 2006, the Centre for Plant Quarantine Pests at the Canadian Food Inspection Agency (CFIA; www.inspection.gc.ca) conducted pheromone trapping surveys of most commercial apple growing areas within Canada. These surveys detected *S. myopaeformis* at several locations in B.C.: near Cawston and Keremeos in the Similkameen Valley, in Oliver,

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Osoyoos and the Ellison suburb of Kelowna in the Okanagan Valley, and near Langley, Abbotsford, and Yarrow in the Fraser Valley (Canadian Food Inspection Agency 2006). One moth catch in Fingal, Ontario, was the only Canadian detection outside B.C. (Beaton and Carter 2006). By 2008, the insect was officially established in areas of Washington State, USA (LaGasa *et al.* 2009), adjacent to the Fraser Valley detections. Tree surveys in 2008 confirmed the species had invaded 97% of all apple orchards within the Similkameen Valley, and in 39% of the affected orchards more than 80% of the trees were infested (Cossentine *et al.* 2013).

Apple clearwing moth (ACM) can be a destructive pest of commercial apple trees, *Malus domestica* Borkhausen (*Rosacea*), particularly when grown on clonal, size-controlling rootstocks in high-density plantings (Dickler 1976). Damage from ACM has increased throughout Eurasia wherever apple industries have implemented this more intensive apple-production technology (Abd Elkader and Zaklama 1971; Dickler 1976; Blaser and Charmillot 1984; Castellari 1987; Balázs *et al.* 1996; Sahinoglou *et al.* 1999; Ateyyat 2006; Kutinkova *et al.* 2006). An identical situation occurred in eastern North America, where the congeneric, native dogwood borer, *Synanthedon scitula* (Harris), became a serious pest after the apple industry converted to high-density plantings (Riedl *et al.* 1985; Kain and Straub 2001).

The heightened pest status of these apple-infesting sesiids has occurred because clonal rootstocks promote formation of adventitious root primordia (burr knots) near the rootstock–scion graft union. These above-ground burr knots are either preferred oviposition sites or susceptible points of entry for neonate larvae (Dickler 1976; Bergh and Leskey 2003). Repeated infestation and feeding by sesiid larvae eventually depletes burr knot tissue, and this leads to feeding in the cambial layer, which ultimately girdles the bark and provides sites for infections that weaken or kill the tree (Dickler 1976; Iren *et al.* 1984; Weires 1986).

Given the European experience with ACM, the potential for severe damage seemed likely after its introduction to the Similkameen Valley. Most apple orchards in this production region had been converted to high-density plantings in the preceding decade (personal observation and unpublished Okanagan Tree Fruit Authority reports), at least 50% of the apples were under organic production (Mullinix 2005), and an area-wide programme controlling codling moth had virtually eliminated use of synthetic insecticides in apples (Judd and Gardiner 2005). An expectation of impending damage stimulated pest management research on several fronts (Judd 2008; Cossentine *et al.* 2010; Aurelian 2011; Eby 2012; Kwon 2013), including chemical controls that are still a primary method of managing many sesiid pests of woody trees and shrubs (Johnson and Lyon 1991).

Traditional insecticidal sprays are effective against most borers only when lethal residues are present on bark during a brief period between egg deposition and initial entry of neonate larvae into the tree (Potter and Timmons 1983). Control of *S. myopaeformis* with many short-lived insecticides is difficult because adults emerge and oviposit from June to August (Judd 2008) and larvae spend the majority of life feeding beneath the bark—up to two years in some areas (Dickler 1976). The extended field life of organochlorines (e.g., endosulfan) and organophosphates (e.g., chlorpyrifos), especially when applied to tree trunks using large volumes of water (drenches), provides levels of efficacy that led to their global use for controlling apple-infesting sesiids for more than four decades (Abd Elkader and Zaklama 1971; Frankenuyzen 1979; Warner and Hay 1985; Balázs *et al.* 1996; Ateyyat 2005). With these insecticidal classes under review or being deregistered in Canada in 2008, largely a result of the 1996 USA *Food Quality Protection Act*, urgent need arose for controls to address the problem facing many Similkameen apple producers, particularly organic producers.

The objectives of this study were: (1) to screen a number of new synthetic insecticides having novel chemistries that purportedly have reduced risks; (2) to evaluate the use of several organically approved spray materials, including live nematodes; and, (3) to compare the efficacy of these various products when applied at different times during the growing season.

MATERIALS AND METHODS

Test Sites. Screening trials for control of ACM with synthetic insecticides and nematodes were

established on two conventionally managed commercial apple orchards in Cawston, B.C. (49.15 N and -119.74 W). Three screening trials in 2008 and one in 2009 were conducted on Farm 1, in a 14-row, 12-year-old, slender-spindle planting of 'Gala' apple cultivar grafted to M9 rootstock (Block 1). One summer 2009 trial was conducted in a six-row, eight-year-old, slender-spindle planting of 'Fuji' apple cultivar grafted on Ottawa-3 rootstock (Block 2), also on Farm-1, about 50 m from Block 1. In both blocks 1 and 2, the alley spacing between tree rows was 3 m, and tree spacing within rows was 0.75–1.0 m.

Two nematode-only screening trials were conducted on Farm 2, located on Barcello Road about two kilometres south of Farm 1, in a mature, double-row, slender-spindle planting of 'Gala' apple cultivar grafted to M26 rootstock (Block 3). The alley and tree spacing in Block 3 was 3×1 m.

Spray Materials and Rates. Three insecticides tested as possible organic chemical control products included: (1) Entrust® 80W (80% spinosad) at 109 g of product ha⁻¹ [Dow AgroSciences, Calgary, Alberta]; (2) Crocker's Fish Oil® [Crocker's Fish Oil Inc., Quincy, Washington State, USA]; and (3) Purespray Green™ Spray Oil 13E [Petro-Canada Lubricants Inc., Mississauga, Ontario]. Both oils were applied as 1% (volume / volume) suspensions in water.

Four synthetic insecticides, each with a novel chemistry and some recently registered for use by conventional apple producers in Canada (British Columbia Ministry of Agriculture 2010), included: (1) Altacor® (35% chlorantraniliprole) at 285 g of product ha⁻¹ [DuPont™ Canada Company, Mississauga, Ontario]; (2) Belt® (39% flubendiamide) at 300 and 350 ml of product ha⁻¹ in 2008 and 2009, respectively [Bayer CropScience, North Carolina, USA]; (3) Delegate™ WG (25% spinetoram) at 420 g of product ha⁻¹ [Dow AgroSciences, Calgary, Alberta]; and, (4) Rimon® 10EC (10% novaluron) at 2.8 L of product ha⁻¹ [Chemtura Canada Company, Elmira, Ontario].

Nematode Products and Rates. Three species of nematodes were tested as possible biological control products for organic apple producers: (1) *Steinernema feltiae* (Filipjev) (1×10⁵ infective juvenile [IJ] nematodes in 100 ml of water / tree [May 2008 source: L. Lacey, USDA, Wapato, USA; October 2008 source: Westgro Sales Inc., Delta, B.C.]), (2) *Heterorhabditis bacteriophora* Poinar (1.5×10⁵ IJ [May 2009] and 5×10⁵ [August 2009] nematodes in 100 ml of water / tree [(Biobest Biological Systems, Leamington, Ontario)], and (3) *Steinernema carpocapsae* (Weiser) 7×10³ IJ [May 2009] and 3.9×10³ IJ nematodes [August 2009] in 100 ml of water / tree [The Bugfactory Ltd., Nanoose Bay, B.C.].

Spray Techniques. Unless noted otherwise, all insecticide sprays were applied using a calibrated, hand-held spray gun (Wheaton Gunjet, Spraying Systems Company, Wheaton, Illinois, USA) equipped with a Teejet D6 nozzle and attached to a truck-mounted sprayer (Rittenhouse, St. Catharines, Ontario) that operated at 200 psi and was calibrated to deliver 2,000 L ha⁻¹. All sprays were applied to the base of individual trees from soil level to a height of the lowest scaffold limbs; in general, this corresponded to the height of the lowest wire on the trellis system and was approximately 50 cm above ground.

Spray Timing. A total of seven screening trials—three in 2008 and four in 2009, including two nematode-only trials—were conducted to evaluate the various control products when applied during three different seasons: (1) spring, (2) summer, and (3) autumn. The spring post-bloom spray timing was considered *curative* and primarily targeted mature larvae as they expelled frass from feeding galleries, prepared cocoons, and pupated near their gallery entrances. Each year, spring applications were made following an examination of all trees in all test blocks (Block 1 on 15 May 2008; Block 2 on 10 May 2009; Block 3 on 21 May 2009). These examinations determined there was a 100% level of tree infestation, based on the presence of larval frass having been expelled from feeding galleries near the rootstock–scion graft unions. This expulsion of frass in spring is commonly observed in B.C. whenever burr knots on dwarfing apple trees are infested by ACM.

The in-season, mid-summer spray timing was considered *preventative*, because it targeted eggs and neonate larvae before they gained entry to the tree. The timings for these preventive summer sprays were based on a preliminary phenology model that attempted to predict 50% and 95% adult emergence (Judd 2008). Phenology of moth emergence was approximated by catches of males in

traps containing the female sex pheromone 3Z,13Z-octadecadienyl acetate (Judd *et al.* 2011). Two all-yellow Unitraps® (AgBio Inc., Westminster, Colorado, USA) were hung at 1.5 m above ground in the middle of each test block. Traps were baited with grey halobutyl rubber septa (West Co., Lyonville, Pennsylvania, USA) that were impregnated with 10 mg of 3Z,13Z-octadecadienyl acetate (Pherobank, Wageningen, The Netherlands; > 95% isomeric purity) dissolved in 200 µL of HPLC grade hexane (Aldrich Chemical Co., Milwaukee, Wisconsin, USA). We placed a 2.5×5.0 cm insecticidal strip containing 10% dichlorvos (Vaportape™ II, Hercon Environmental, Emigsville, Pennsylvania, USA) inside the bucket of each composite Unitrap to improve capture (Judd and Eby 2014). Traps were checked weekly, and all apple clearwing moths were counted and removed. A single pheromone lure lasted the entire season.

Hourly air temperatures throughout the study were recorded at a centrally located orchard in Cawston, B.C., from 1 January through 31 December each year. Temperature readings were made using a HOBO® data logger (Onset, USA) housed in a 1-m high Stevenson screen. Daily degree-day (DD) summations above an arbitrary 10° C developmental base temperature (DD_{10°C}) and below a 31° C upper developmental threshold were calculated by fitting a sine wave (Allen 1976; case 4) to daily air temperature minima and maxima using the computer program described by Higley *et al.* (1986).

The late-summer and early-autumn post-harvest sprays targeted late-season hatching eggs and larvae that may not have gained deep entry beneath the bark. All post-harvest sprays were applied after catches of male moths in traps had ceased.

Experimental Trials and Assessments. In Experiments 1 to 5, all treatments were assigned to test plots using a randomised complete block design with 4 to 10 replicates depending on the trial. Each replicate block consisted of a single 6-metre-long tree-row containing 7 to 9 trees, depending on the exact tree spacing. Data in experiments 1 to 5 were collected only from the five central trees in each test block. One to two trees on either end of each 6-m row served as guard trees to separate adjacent treatments.

Experiment 1 (2008) had five replicates and evaluated single spring applications of Altacor, Belt, Delegate, Rimon, Entrust, Crocker's Fish Oil, a nematode treatment, and water-only control. All treatments were applied on 27 May 2008 (Fig. 1). The nematode treatment in this experiment contained *S. feltiae* and was applied with a backpack sprayer at a rate of 1×10^5 IJ nematodes in 100 ml of water / tree. After applying the nematode treatment, a water-soaked piece of burlap was wrapped around the treated rootstock–scion graft union of each tree and then covered with wet cardboard that was stapled in place. The purpose of wraps was to slow desiccation and promote nematode efficacy (Lacey *et al.* 2010; Shapiro-Ilan *et al.* 2010; Cottrell *et al.* 2011).

The efficacy of the spring 2008 treatments in Experiment 1 was assessed on 5 August 2008 and again on 13 August 2009. Efficacy was measured by counting the numbers of pupal exuviae that protruded from the bark within a 25-cm zone from the ground up. This zone usually included the rootstock–scion graft union. Pupal exuviae are evidence of adult emergence and often remain attached to the tree for several weeks to months.

Experiment 2 (2008) had 10 replicates and evaluated two summer applications of Altacor, Belt, Delegate, Rimon, Entrust, and Crocker's Fish Oil, compared to a water-only control. The two summer sprays were applied on the 8 and 22 July 2008 (Fig. 1). The nematode treatment was excluded from Experiment 2, because it was assumed that extreme summer temperatures would eliminate its efficacy. Pupal exuviae were counted and removed from all test trees with fine forceps on 5 August 2008. This count confirmed the level and uniformity of the infestation that must have existed before summer treatments were applied, because these pupal exuviae would have arisen from larvae feeding in the tree since 2007, well before our controls were applied. The efficacy of the summer 2008 treatments in Experiment 2 was assessed by counting pupal exuviae on 13 August 2009.

Experiment 3 (2008) had five replicates and evaluated single autumn applications of Altacor, Belt, Delegate, Rimon, Entrust, and a nematode treatment, compared to a water-only control. The nematode treatment in this experiment also contained *S. feltiae* applied with 1×10^5 IJ nematodes in 100 ml of water / tree, but unlike the spring nematode treatment (Experiment 1), nematodes were not covered with burlap or cardboard to prevent desiccation. As before, all pupal exuviae

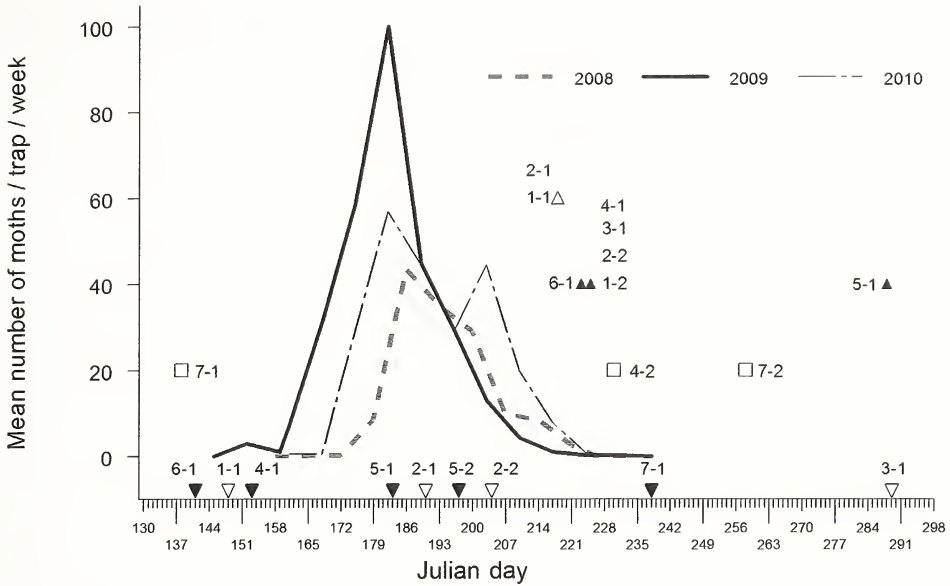


Figure 1. Treatment application dates in each of three 2008 experiments (open inverted triangles with associated experiment - spray numbers) and four in 2009 (solid inverted triangles with associated numbers), with follow-up population assessments in 2008 (open triangles with associated experiment - assessment numbers), 2009 (solid triangles with associated numbers), and 2010 (open squares with associated numbers), in relation to seasonal catches of male *S. myopaeformis* in pheromone-baited all-yellow Unitraps® at Cawston B.C., 2008–2010.

were counted and removed from test trees on 5 August 2008. These counts served as a pre-treatment assessment to ensure the levels of infestation were uniform across all treatments and replicates. All treatments in Experiment 3 were applied on 15 October 2008, and their efficacy was assessed as before on 13 August 2009.

Experiment 4 (2009) had four replicates and evaluated single spring applications of Altacor, Belt, Delegate, Rimon, Purespray Green Oil, Belt *plus* Purespray Oil, and a Rimon *plus* Purespray Oil treatment, compared to a water-only control. The Belt *plus* oil and Rimon *plus* oil treatments were tested using the same insecticide product rates as used in Experiment 1, but materials were tank-mixed with the 1% oil in water suspension (volume to volume). All treatments in Experiment 4 were applied on 2 June 2009, and their efficacy was assessed on 13 August 2009 and again on 18 August 2010.

Experiment 5 (2009) had five replicates and evaluated two summer applications of Altacor, Belt, Delegate, Rimon, Entrust, and Purespray Green Oil, compared to a water-only control. The two summer sprays were applied on 2 and 16 July 2009 (Fig. 1). On 13 August 2009, we counted and removed all the pupal exuviae from our test trees as we had done in Experiment 2. This provided an assessment of the level of infestation that existed before treatments were applied.

Our original intention was to assess the efficacy of sprays applied in Experiment 5 one year after treatment, in August 2010, as we had done in Experiment 2. However, in August 2009, the grower informed us that he intended to remove our test block in October 2009. We switched our assessment method and took the opportunity to examine the harvested trees and count the numbers of larvae using a destructive sample. Two test trees in the centre of each treatment block were cut 30 cm above ground and pulled from the ground with an excavator, exposing the roots. All roots more than 30 cm from the tree trunk were removed with pruning shears. Pruned tree sections were returned to the laboratory, where they were washed with a low-pressure water hose. All bark 25 cm above and below ground level was removed with a knife, and exposed larvae were counted and classified as being found above or below ground. Chilled larvae were measured with

a small ruler and classified as either ≤ 9 mm or > 9 mm. The smallest larvae were expected to have arisen from eggs laid in 2009; thus, their reduction should represent the impact of summer 2009 treatments. The largest larvae may have arisen from oviposition in 2008 before our treatments were applied.

Nematode plus Barrier Experiments. These nematode-only experiments were conducted in Block 3 located on Farm 2, about 2 km south of Farm 1. Experiment 6 (2009) used a split-plot design with five replicate blocks to evaluate the efficacy of two different species of nematode when applied to tree trunks as a single spring application, alone or in combination with different barrier wraps that help to prevent desiccation of nematodes (Lacey *et al.* 2010; Shapiro-Ilan *et al.* 2010; Cottrell *et al.* 2011). On 21 May 2009, two nematode treatments, *S. carpocapsae* (7×10^3 IJ nematodes [Sc] in 100 ml of water / tree) and *H. bacteriophora* (1.5×10^5 IJ nematodes [Hb] in 100 ml of water / tree), and a 100-ml water-only control were each applied to 30 different trees within each of five replicate blocks. All sprays were applied with a calibrated backpack sprayer. Air temperature during application was 15°C but reached $>19^\circ\text{C}$ on the day of application. Immediately following these three applications, each group of 30 trees was further subdivided into three groups of 10 trees. One group of 10 nematode- or water-treated tree trunks was wrapped with a sawdust mixture (0.8 L of sawdust: 0.55 L vermiculite : 0.4 L perlite : 10 ml white glue [Elmer's white] : 1 L of tap water) that was moulded onto trees by hand (SDW). A second group of 10 treated trees was wrapped with wet cardboard that was stapled in place (CBW). The third group of 10 treated trees was left unwrapped (UW). Thus, there were nine nematode-wrap treatment combinations in total: (1) Sc-SDW, (2) Sc-CBW, (3) Sc-UW, (4) Hb-SDW, (5) Hb-CBW, (6) Hb-UW, (7) water-SDW, (8) water-CBW, and (9) water-UW. All CBW were removed on 5 June 2009. The SDW were left in place to erode naturally from the effects of irrigation and rain, but all remaining sawdust material was completely removed on 11 August 2009. The original plan was to use higher and comparable numbers of nematodes in each nematode treatment; however, laboratory counts of live nematodes were lower than the estimated counts on labelled commercial packages. Nematode infectivity for each treatment was confirmed in the laboratory using infection rates on third-instar oblique-banded leafroller, *Choristoneura rosaceana* (Harris). The efficacy of nematode and barrier treatments (SDW and CBW) was subsequently measured by counting numbers of empty pupal exuviae that protruded from the bark within a 25-cm zone from the ground up. Counts of pupal exuviae were made on all trees on 11 August 2009 (Fig. 1).

Experiment 7 (2009) was also conducted in Block 3 on Farm 2, but used a different set of trees than in Experiment 6. Experiment 7 employed a split-plot experimental design with five replicates to evaluate the efficacy of a single late-summer application of two different species of nematode when applied to tree trunks, alone or in combination with the SDW. On 26 August 2009, two nematode treatments, *S. carpocapsae* (3.9×10^3 IJ nematodes [Sc] in 100 ml of water / tree) and *H. bacteriophora* (5×10^5 IJ nematodes [Hb] in 100 ml of water / tree), and a 100 ml water-only control were each applied using a calibrated backpack sprayer to the bottom 45 cm of each of 20 trees within each of five replicate blocks. Immediately after the nematode treatments were applied, each group of 20 trees was subdivided into two groups of 10 trees. One group of 10 nematode- or water-treated tree trunks was left uncovered (Sc-UW, Hb-UW, and Water-UW, respectively), and the other groups of 10 treated trees were wrapped with a SDW as described in Experiment 6 (Sc-SDW, Hb-SDW and Water-SDW, respectively). Air temperature was above 15°C at the time all nematode treatments were applied, but reached 27°C later that day. Any SDW still remaining on trees on 18 May 2010 was removed before any moths emerged (Fig. 1). The efficacy of the August 2009 nematode treatments was measured by counting pupal exuviae protruding from the trunks and on the soil around all treated trees on 29 September 2010.

Statistical analyses. Pupal count data generated in Experiments 1 through 5 were subjected to two-way randomised block analyses of variance (ANOVA), with repeated measures in 2008 and 2009 (Experiments 1 through 3), or in 2009 and 2010 (Experiment 4). Larval count data from harvested trees in Experiment 5 were analyzed by a two-way randomised block ANOVA. Mean numbers of pupal exuviae (Experiments 1 through 4) or mean numbers of larvae (Experiment 5) in treatment trees were compared to counts from control trees using the *post hoc* Dunnett test and an experiment-wise error rate set at $\alpha = 0.05$ (Zar 1984). All insect count data were tested for

normality (Kolmogorov-Smirnov Test) and equality of variances (Levine's Median Test) to ensure they met the assumptions of the ANOVA. All of these analyses were performed using SigmaPlot®-12 (Systat Software Inc., San Jose, California, USA).

In Experiments 6 and 7, the effects of nematode treatments and tree trunk wraps were tested using a two-way split-plot ANOVA, with replication and barriers as the main effects (SAS 2008). Mean numbers of pupal exuviae in each treatment were compared *post hoc* using Tukey's HSD test and an experiment-wise error rate set with $\alpha = 0.05$.

RESULTS and DISCUSSION

Seasonal pheromone trap catches indicate that ACM had a single flight period that started in late-May and culminated by mid-August during 2008 – 2010 in Cawston, B.C. (Fig. 1). By overlaying our experimental spray dates on catch curves, we were able to compare spray timings in various experiments with seasonal phenology of ACM. In 2008, spring treatments in Experiment 1 were applied at 201 DD_{10°C} from 1 January 2008, almost three weeks before the first moth was caught (Fig. 1)—meaning many larvae had likely not yet formed cocoons or pupated. Among the treatments applied in spring 2008, Rimon, Belt, and nematodes all caused a significant reduction in emergence of moths in the summer of 2008, relative to emergence from the control trees (Table 1). This indicates that a single spring application of these materials can be curative in action, because they kill mature larvae in their feeding galleries or exit holes before pupation.

A single spring application of Rimon may contribute to mortality over two seasons because trees treated with Rimon in spring 2008 had a statistically significant year-over-year reduction (–66.7%) in pupal exuviae counts in 2009 that was more than twice that seen in water controls (Table 1). We are uncertain whether ACM has a one- or two-year lifecycle in B.C. (Judd 2008); therefore, it remains unclear whether the reduced adult emergence in 2009 from trees treated in 2008 (Table 1) is the result of a curative action in spring 2008 or a carry-over preventative action in summer 2008. If ACM has a one-year life cycle and Rimon has a long residual period, the product could prevent further infestation by killing eggs and neonate larvae appearing on trees in summer 2008. However, if ACM has a two-year lifecycle, then immature larvae present in spring 2008 but destined to emerge in 2009 could have been killed by Rimon through a curative action. Nothing precludes both things happening. Either way, control of ACM with Rimon applied in spring appears promising. Adult emergence from trees treated with Belt in spring 2008 was significantly different than the control trees in summer 2009 (Table 1), but there was no significant year-over-year reduction from the spring Belt treatment—a reduction similar to that seen on the control trees was evident (Table 1).

Summer 2008 treatments in Experiment 2 were applied at 579 and 764 DD_{10°C} after 1 January 2008, which corresponded to 51% and 95% cumulative moth catch, respectively (Fig. 1). Among the insecticide treatments applied twice in summer 2008, Altacor, Belt, Delegate, Entrust, and Rimon all caused significant reductions in adult emergence in 2009 (Table 1). The summer 2008 Rimon treatment exhibited the greatest year-over-year reduction (–96.4%) in pupal exuviae counts (Table 1).

Autumn 2008 treatments in Experiment 3 were applied at 1445 DD_{10°C} after 1 January 2008, about seven weeks after the last moths were caught (Fig. 1). Rimon was the only treatment applied once in autumn 2008 (Experiment 3) that caused a significant reduction in adult emergence in 2009 compared with emergence from the control trees (Table 1). Pupal counts on Rimon-treated trees in 2009 were 89.2% lower than the pre-test counts made in summer 2008 (Table 1). It should be noted that several autumn treatments in Experiment 3, including the control, exhibited significant year-over-year reduction in pupal counts (Table 1). These generalized reductions may indicate that eggs and neonate larvae can be damaged or dislodged by high-pressure or high-volume handgun sprays, but lack of a non-treatment control prevents us from making definitive conclusions on these year-over-year results.

Spring treatments in Experiment 4 were applied on 2 June 2009, with an accumulation of 288 DD_{10°C} after 1 January 2009, and almost one week after the first moth was caught (Fig. 1). This means that many of the late-instar larvae this spray timing was targeting in 2009 had accumulated

Table 1

Control of apple clearwing moth with reduced-risk insecticides and nematodes when applied as spring post-bloom and autumn post-harvest *curative* treatments or as summer *preventative* treatments in Cawston, B.C., Canada, in 2008.

Experiment no. and replicates with treatment timings	Treatments ¹	Product rate ha ⁻¹	Mean (± SE) number of pupal exuviae / tree / assessment year ²		Percent change in exuviae counts year-to-year ³
			2008	2009	
Exp. 1 (n = 5) 27 May 2008	Water control	2000 L	2.43 ± 0.49 a	1.81 ± 0.29 a	– 25.5
	Crocker's Fish Oil	20 L	1.78 ± 0.17 a	1.61 ± 0.24 a	– 9.6
	Nematodes	105 IJs	1.17 ± 0.23 b	1.80 ± 0.34 a	+ 53.8
	Altacor	285 g	1.73 ± 0.32 a	1.05 ± 0.16 a	– 39.3
	Belt	300 mL	1.08 ± 0.13 b	0.83 ± 0.18 b	– 23.1
	Delegate	420 g	1.59 ± 0.31 a	1.57 ± 0.15 a	– 1.3
	Entrust	109 g	2.05 ± 0.43 a	1.51 ± 0.27 a	– 26.3
	Rimon	2.8 L	0.71 ± 0.13 b	0.06 ± 0.03 b	– 66.7*
Exp. 2 (n = 10) 8, 22 July 2008	Water control	2000 L	2.58 ± 0.32 a	2.16 ± 0.21 a	– 15.4
	Crocker's Fish Oil	20 L	1.86 ± 0.27 a	2.14 ± 0.32 a	+ 10.5
	Altacor	285 g	2.42 ± 0.45 a	1.22 ± 0.27 b	– 50.0*
	Belt	300 mL	2.61 ± 0.37 a	0.62 ± 0.17 b	– 76.9*
	Delegate	420 g	2.27 ± 0.32 a	1.09 ± 0.11 b	– 52.2*
	Entrust	109 g	2.69 ± 0.48 a	1.35 ± 0.26 b	– 51.8*
	Rimon	2.8 L	2.78 ± 0.41 a	0.13 ± 0.05 b	– 96.4*
Exp. 3 (n = 5) 15 October 2008	Water control	2000 L	4.13 ± 0.64 a	1.90 ± 0.41 a	– 53.9*
	Nematodes	10 ⁵ IJs	2.93 ± 0.51 a	1.23 ± 0.13 a	– 58.0*
	Altacor	285 g	3.14 ± 0.64 a	1.84 ± 0.59 a	– 41.4
	Belt	300 mL	3.80 ± 0.32 a	1.20 ± 0.22 a	– 68.4*
	Delegate	420 g	3.57 ± 0.47 a	1.65 ± 0.37 a	– 53.8*
	Entrust	109 g	2.93 ± 0.38 a	2.05 ± 0.39 a	– 30.0
	Rimon	2.8 L	3.53 ± 0.24 a	0.38 ± 0.16 b	– 89.2*

¹ All treatments except nematodes were applied using a handgun and delivered in water at a rate of 2000 L ha⁻¹ to each rootstock scion-graft union. All *Steinernema feltiae* nematode treatments (IJs = infective juvenile nematodes) were applied using a back sprayer and delivered in 100 ml of water.

² Treatment means within a column for each experiment followed by the same letter as the control group are not significantly different (Dunnnett's test, $\alpha = 0.05$) following significant randomised block ANOVA ($P < 0.05$), with repeated measures in 2008 and 2009.

³ Percentages followed by an asterisk (*) denote significant change in counts between yearly assessments based on two-tailed paired *t*-tests, $P < 0.05$.

87 DD_{10°C} more than the same larval stages in 2008. Many of these late-instar larvae had probably cocooned or pupated at the time this spray was applied. This late spray timing likely explains why none of the spring 2009 treatments in Experiment 4, including Rimon, had any impact on adult emergence in summer 2009 (Table 2). Rimon is an insect-growth regulator that must be absorbed by eggs or ingested by larvae to be effective. Its primary mode of action is disruption of cuticle formation and deposition when insects change from one developmental stage to another, resulting in death at moulting. Due to this mode of action, a late-spring Rimon treatment in Experiment 4 would have no effect on insects that have cocooned, completed moulting, or pupated. However, this late-spring 2009 Rimon treatment did have an impact on emergence in 2010 (Table 2). Once

Table 2

Control of apple clearwing moth with reduced-risk insecticides when applied once as spring post-bloom curative treatments in Cawston, B.C., Canada, in 2009.

Experiment no. and replicates with treatment timings	Treatments ¹	Product rate ha ⁻¹	Mean (± SE) number of pupal exuviae / tree / assessment year ²		Percent change in exuviae counts year-to-year ³
			2009	2010	
Exp. 4 (n = 4)	Water	2000 L	3.20 ± 0.97 a	3.85 ± 1.87 a	+ 20.3
2 June 2009	Oil	20 L	2.50 ± 0.57 a	3.15 ± 1.45 a	+ 26.0
	Altacor	285 g	3.50 ± 1.60 a	2.80 ± 1.28 a	– 20.0
	Belt	350 ml	2.30 ± 0.46 a	1.60 ± 0.37 a	– 30.4
	Belt plus Oil	350 ml + 20 L	2.30 ± 0.88 a	0.55 ± 0.26 b	– 76.1*
	Delegate	420 g	1.40 ± 0.32 a	2.00 ± 1.23 a	+ 42.9
	Rimon	2.8 L	2.40 ± 1.43 a	0.50 ± 0.26 b	– 79.2*
	Rimon plus Oil	2.8 L + 20 L	2.35 ± 0.79 a	0.60 ± 0.45 b	– 74.5*

¹ All treatments were applied using a handgun and delivered in water at a rate of 2000 L ha⁻¹ to each rootstock scion-graft union. Oil is Purespray Green.
² Treatment means within a column followed by the same letter as the control group are not significantly different (Dunnett's test, $\alpha = 0.05$) following significant ($P < 0.05$) randomised block ANOVA with repeated measures in 2009 and 2010.
³ Percentages followed by an asterisk (*) denote significant change in counts between yearly assessments based on two-tailed paired *t*-tests, $P < 0.05$.

again, the effects of Rimon that manifest in 2010 could be due to a curative or preventative action. Late-spring applications of Rimon could control populations of ACM by killing immature larvae already in the tree or by killing eggs and neonate larvae appearing on trees in summer 2009. Apparently, Altacor, Belt, and Delegate either do not penetrate tree bark sufficiently to reach immature larvae or their residual activity is significantly less than Rimon, as none of these insecticides when applied in late-spring 2009 had any effect on emergence of adults in summer 2010 (Table 2). The addition of oil to an application of Belt did appear to increase its efficacy, because the Belt *plus* Oil treatment applied in late-spring 2009 caused a significant reduction in adult emergence in summer 2010 (Table 2). The Rimon *plus* Oil treatment was no more effective than Rimon applied alone (Table 2).

Summer 2009 treatments in Experiment 5 were applied at 583 and 759 DD_{10°C} after 1 January 2009, corresponding to 71% and 94% cumulative moth catch in 2009, respectively (Fig. 1). Given the grower's decision to remove this test block in autumn 2009, we were forced to assess the impact of our summer 2009 sprays before they would likely show their greatest efficacy. Sprays applied in summer likely have their greatest impact on ACM populations by killing eggs and neonate larvae before they enter the bark. On trees harvested in autumn 2009, we expected to see the greatest impact of summer 2009 sprays on a younger, smaller-size class of larvae. Rimon was the only insecticide treatment in summer 2009 that caused a significant reduction in larval counts compared with those from control trees (Table 3). Whether we compared counts of larvae found on above- or below-ground parts of trees, the effect of Rimon was greatest on the smallest larval class (Table 3). We were somewhat surprised to find ACM larvae on below-ground tree parts, as we have found no literature reference to this species being subterranean. The occurrence of subterranean larvae may be because the densities of this invasive species are much greater in the Similkameen Valley than anywhere in its natural range (Špatenka *et al.* 1999).

Nematode plus Barrier Experiments. Although effective in one spring 2008 trial (Table 1), nematodes had little impact on controlling larval populations of ACM. Efforts to improve their efficacy focused on preventing desiccation at the time of application. Spring 2009 applications of nematodes in Experiment 6 were applied at 157 DD_{10°C} after 1 January 2009, three weeks before the first moths were caught (Fig. 1) and well before larvae had pupated. At this spray timing, neither the *H. bacteriophora* nor the *S. carpocapsae* nematode treatments appeared effective in suppressing the number of ACM that emerged from treated trees in 2009 (Fig. 2). These results

Table 3

Control of apple clearwing moth with reduced-risk insecticides when applied as summer preventative treatments in Cawston, B.C., Canada, 2009 (Exp. 5, n=5).

Treatment ¹	Product rate ha ⁻¹	Mean (±SE) number of pupal exuviae / tree before harvest ²	Mean (±SE) number of larvae in different size classes and parts of harvested trees ³				Mean total number of larvae / tree
			0-25 cm above ground		0-25 cm below ground		
			Larva length		Larva length		
			≤9 mm	>9 mm	≤9 mm	>9 mm	
Water	2000 L	4.6 ± 1.1 a	10.2 ± 2.1 a	2.8 ± 0.5 a	1.5 ± 0.5 a	0.4 ± 0.2 a	14.9 ± 2.5 a
Oil	20 L	4.5 ± 1.2 a	9.7 ± 1.7 a	4.5 ± 0.9 a	1.4 ± 0.4 a	0.8 ± 0.4 a	16.4 ± 1.6 a
Altacor	285 g	4.0 ± 0.9 a	5.2 ± 0.9 a	3.0 ± 0.6 a	1.9 ± 0.7 a	1.1 ± 0.5 a	11.2 ± 1.6 a
Belt	300 mL	5.9 ± 1.2 a	5.5 ± 1.8 a	3.9 ± 1.2 a	2.8 ± 1.4 a	1.1 ± 0.3 a	12.5 ± 3.1 a
Delegate	420 g	4.6 ± 0.4 a	5.0 ± 1.0 a	3.9 ± 0.9 a	0.4 ± 0.4 a	0.7 ± 0.3 a	10.0 ± 1.7 a
Entrust	109 g	4.2 ± 0.6 a	9.4 ± 2.1 a	3.9 ± 0.9 a	0.3 ± 0.2 a	0.4 ± 0.3 a	13.8 ± 2.6 a
Rimon	2.8 L	4.5 ± 1.5 a	2.9 ± 0.9 b	1.0 ± 0.3 b	0.0 ± 0.0 b	0.0 ± 0.0 a	4.0 ± 1.1 b

¹ All treatments were applied on 2 July and again on 16 July 2009 using a handgun and delivered in water at a rate of 2000 L ha⁻¹ to each rootstock scion-graft union.

² Mean pupal exuviae counts within this column were not significantly different by randomised block ANOVA ($F_{6, 24} = 0.326, P = 0.918$).

³ Treatment means within a column followed by the same letter as the control group are not significantly different (Dunnnett's test, $\alpha = 0.05$) following significant randomised block ANOVAs ($P < 0.05$). Two trees were harvested from each treatment replicate on 15 October 2009.

could be due, in part, to a low number of IJ nematodes in each of these commercial nematode products: there were large numbers of dead nematodes in both suspended products.

The efficacy of commercial nematode treatments applied in spring 2009 was not impacted by wrapping tree trunks in wet cardboard (CBW) for one week after nematode application (Fig. 2), but was improved by the SDW that remained in place from May to August. Trees receiving the SDW had significantly less ACM emergence than UW or CBW trees ($F_{2, 429} = 39.7; P < 0.0001$). The effect of the SDW appears independent of the nematode treatment because significantly fewer pupal exuviae were found on water-treated control trees receiving the SDW than on water-treated trees that received no wrap (see UW, Fig. 2). Although the SDW deteriorated over time, especially on trees hit by irrigation water, much remained in August and a significant inverse correlation existed between the percentage of the sawdust barrier remaining at the end of the adult emergence period and the number of pupal exuviae on each tree ($n = 192; r = -0.34, P < 0.0001$). It is important to note that any impact the SDW might have had on oviposition during summer 2009 would not have contributed to the smaller number of pupal exuviae counted in autumn 2009, because pupae arising from 2009 eggs would not appear until 2010 at the earliest.

The August 2009 application of nematodes with and without SDW in Experiment 7 was made at 1303 DD_{10°C} after 1 January 2009, at least one week after the last moths were caught in 2009 (Fig. 1). The SDW, which remained in place from 26 August 2009 to 18 May 2010, was removed well before moths emerged in 2010 (Fig. 1). August 2009 nematode treatments without SDW appeared to have no effect on the emergence of ACM in summer 2010 (Fig. 3), whereas the number of ACM emerging from trees that received nematodes with SDW was significantly lower ($F_{1, 285} = 25.31; P < 0.0001$) than the number emerging from trees left unwrapped over the winter months (Fig. 3). Again, a significant inverse correlation was found between the percentage of the

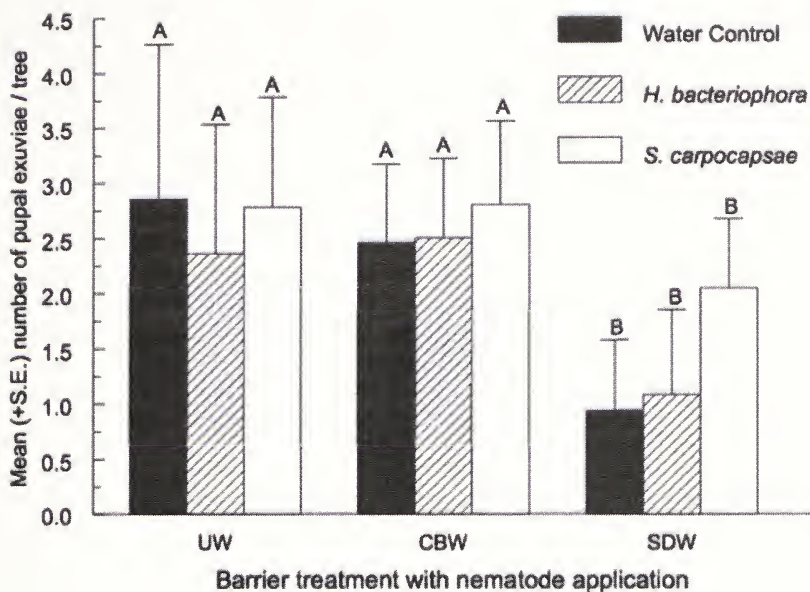


Figure 2. Mean numbers of ACM pupal exuviae recovered from tree trunks on 11 August 2009 after applying water or two nematode treatments, *Heterorhabditis bacteriophora* and *Steinernema carpocapsae*, on 21 May 2009 and left unwrapped (UW), wrapped with cardboard (CBW) for one week, or wrapped with a sawdust mixture (SDW) from 21 May 2009 to 11 August 2009 after application. A significant ($F_{2, 429} = 39.7$; $P < 0.0001$) difference between barrier treatments is indicated by different letter superscripts above bars. There were no significant treatment effects within each barrier treatment ($P > 0.05$).

SDW that remained in May—before ACM emergence had begun (Fig. 1)—and the number of pupal exuviae recovered from each tree in late summer 2010 ($n = 192$, $r = -0.24$, $P = 0.0006$).

Although the ANOVA indicated that nematode treatments applied in autumn 2009 did not have a significant ($F_{2, 285} = 0.52$; $P = 0.5949$) independent impact on the number of ACM emerging in 2010, there was a significant wrap \times treatment interaction ($F_{2, 285} = 4.98$; $P = 0.0075$) that affected the number ACM that emerged. When the data were partitioned and only those trees receiving SDW were compared, treatment with both nematode species was found to significantly ($F_{2, 142} = 5.39$; $P = 0.0056$) lower the number of ACM that emerged compared to the control group (Fig. 3). This result is promising and should be followed up, as it suggests nematodes applied in autumn could be efficacious if combined with the correct physical barrier. The temperature maximum on the day nematodes were applied in August 2009 was higher than the maximum in spring 2009; this may have increased the ability of both nematode species to find and infect hosts in the August study (Fig. 3). It also seems possible that the SDW increases survival of nematodes and/or affects movement of ACM larvae in their feeding tunnels, both of which may increase the efficacy of entomopathogenic nematodes.

ACM is becoming a pest of increasing importance to the B.C. apple industry, especially as the industry becomes economically dependent on replanting new varieties on dwarfing root stocks using high-density planting systems with relatively short rotation times. Our studies suggest conventional apple producers have more options for managing ACM than organic producers as this invasive pest spreads through B.C. (Cossentine *et al.* 20103). Rimon 10 EC appears to be an effective control product for ACM, and its mode of action appears to provide apple producers with varied and flexible spray timing. Spring or autumn applications outside the fruiting season may be the best times to apply Rimon, because these likely have the least impact on beneficial organisms. There are some unpublished reports that Rimon can lead to mite outbreaks if used during the summer months. The most interesting aspect about using Rimon outside the fruiting season is that,

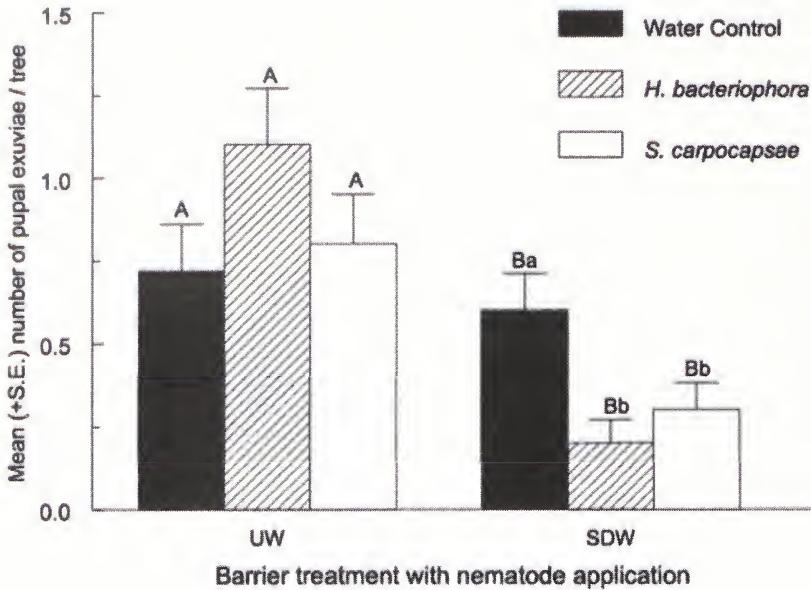


Figure 3. Mean numbers of ACM pupal exuviae recovered from tree trunks on 29 September 2010, after applying water or two nematode treatments, *Heterorhabditis bacteriophora* and *Steinernema carpocapsae*, on 26 August 2009 and left unwrapped (UW) or wrapped with a sawdust mixture (SDW) from 26 August 2009 to 18 May 2010 after application. Bars above each barrier treatment label having different uppercase letter superscripts are significantly different ($F_{1, 285} = 25.31$; $P < 0.0001$), and bars within the SDW barrier treatment with lower-case letter superscripts are significantly different (Tukey's HSD test $\alpha = 0.05$).

when used at these times, it appears to be curative in action and effective against cohorts from two seasons.

Several other insecticides show promise for control of ACM when applied as preventative sprays in summer; some of these are already registered on apples in Canada (Altacor and Delegate). Summer sprays may even be more efficacious when more is known about the seasonal phenology of female ACM and their oviposition patterns in relation to temperature summations. It should be noted that the efficacy of Rimon and other insecticides described in these trials is based on tree-trunk sprays using a handgun with high pressure and high volumes of water. This application method makes sense while infestations of ACM remain restricted to the rootstock graft-union regions of apple trees. Similar results cannot necessarily be expected when ACM infests other parts of the tree or when insecticides are applied as dilute or low-volume sprays typical of air-blast spray techniques used against leafrollers or codling moth (British Columbia Ministry of Agriculture 2010). Recent observations in the Similkameen Valley, where populations of ACM are most extreme (Cossentine *et al.* 2013), have revealed that the pest is beginning to infest limbs higher in the tree, top-grafted scion–tree-trunk unions, and is even entering and killing pruned tree leaders as producers attempt to manage tree height (GJ personal observations). More research will be needed to determine if ACM can be controlled effectively with any insecticides when applied using an air-blast sprayer to these aerial tree limbs.

Our screening trials did not reveal many effective organic options for controlling ACM. Oils and Entrust have become mainstays of integrated pest management for organic apple production in B.C. (British Columbia Ministry of Agriculture 2010), but when used as described herein, these materials had limited impact on infestations of ACM (Tables 1, 3). Some organic apple producers have resorted to applying as many as six trunk sprays of Entrust annually for suppression of ACM (GJ personal observation). This approach is likely uneconomical and unsustainable in the long term, because a lack of alternative insecticides with which to rotate use of Entrust means resistance may develop, not to mention, the many adverse effects Entrust has on important

parasitoids (Williams *et al.* 2003) and predators like lacewings and European earwigs (Mandour 2009; Shaw 2010).

The general ineffectiveness of nematodes in the absence of barriers is disappointing because this would seem to be an excellent organic approach. More research is needed to determine if there are conditions under which nematodes could be made more efficacious. Suboptimal application temperatures and desiccation in dry environments may make nematodes impractical for use in the interior of B.C., but infestations in wetter, cooler coastal areas (Cossentine *et al.* 2013) may be controlled by nematodes.

Although we did not set out to test the efficacy of physical barriers as a method to control ACM, their use in combination with nematodes did lead to some interesting observations. The SDW appeared to be particularly effective at reducing emergence of ACM, especially when applied in spring (Fig. 2). Other studies have found that physical barriers can significantly reduce infestations of *Synanthedon* species. Ateyyat and Al-Antary (2006) found that mounding soil over apple rootstock-scion graft unions or wrapping tree trunks in cheese cloth prevented subsequent adult emergence. Kain *et al.* (2010) used polyethylene fabric, veterinary gauze, and sprays of ethylene vinyl acetate to prevent dogwood-borer infestations. The reduction in emergence of ACM from trees receiving a SDW in spring was not the result of any oviposition-deterrent effect, because the emergent adults were already larvae within the trees before the barrier was applied. It is possible some pre-pupal ACM larvae were deterred from chewing through the SDW, but some pupae did successfully exit through the SDW. In an unpublished laboratory study (JC personal observation), the presence of a SDW for seven weeks did not significantly reduce emergence of ACM adults from infested tree trunks that had been harvested and wrapped compared with those that had not been wrapped. It is possible that the SDW modified larval development time. If SDW insulated tree trunks, it may have delayed ACM development to the point where we may not have seen emergence during a single test season. Unfortunately, the same trees were not re-examined the following year to test this hypothesis.

In conclusion, if infestations of ACM remain restricted to the graft-union areas of dwarfing apples trees, then our research suggests use of physical barriers for control of ACM populations in organic orchards warrants further study. A better understanding of the barriers' mode of action, either as oviposition deterrents or as physical deterrents to larval development and pupal formation or exit from trees, should be sought. With ACM now infesting multiple locations on apple trees, particularly pruned apical stems (GJ personal observations), use of physical barriers and even tree-trunk sprays of Rimon may be limited to situations where populations have not grown to high levels and are restricted to rootstock graft unions. It seems clear from this study that a search for alternative organic methods of controlling ACM is warranted.

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SCIENTIFIC NOTE

**Ambulatory responses of *Laricobius nigrinus*
(Coleoptera:Derodontidae), a hemlock woolly adelgid predator, to
odors from prey, host foliage, and feeding conspecifics**

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Behavioral interactions between insects and their environments are often mediated by volatile cues. Plant-produced chemical cues induced by herbivore activity are often more effective at attracting predators than are cues produced by the herbivore alone (Dicke and van Loon 2000). The presence of herbivore-induced plant volatiles makes foraging by predators more efficient than undirected hunting (Dannon *et al.* 2010), presumably because they indicate the presence of prey. Just as it is common for predators to find prey using olfactory cues, they can also use chemical cues to avoid intraguild competition or aggression (Janssen *et al.* 1995; Cakmak *et al.* 2006, Flowers *et al.* 2007), and evidence suggests that avoidance is a common response to the reception of a conspecific cue from a particular location (Janssen *et al.* 1995; Stout and Goulson 2001; Gnanvossou *et al.* 2003). Understanding these interactions may be important to the implementation of a biological control program in which a natural enemy complex is released and established, because they are integral to how predators find their hosts (Flowers *et al.* 2007). Information about predator interactions can inform the optimal release density of agents on the landscape and increase the efficiency and effectiveness of programs.

Classical biological control practices are being implemented in the eastern United States (U.S.A.) to control on invasive hemlock woolly adelgid (Hemiptera:Adelgidae, *Adelges tsugae*) populations on eastern hemlock (*Tsuga canadensis* (L.) Carriere) and Carolina hemlock (*Tsuga caroliniana* Engelmann) (DeBach 1974; McEvoy 1996; McDonald 2010). *Laricobius nigrinus* Fender (Coleoptera: Derodontidae) is an adelgid predator associated with hemlock woolly adelgid on western hemlock (*Tsuga heterophylla* (Raf.) Sarg.) in the northwestern U.S.A. (Kohler *et al.* 2008) and British Columbia, Canada (Zilahi-Balogh *et al.* 2002). This predator is considered a promising candidate agent for biological control of hemlock woolly adelgid on eastern hemlock (Mausel *et al.* 2012). The objective of this study was to determine whether field-collected *L. nigrinus* responded to host and prey odors in an olfactometer, and to observe whether adding a conspecific individual feeding on hemlock woolly adelgid on host foliage would alter the orientation preferences of *L. nigrinus*.

In 2011 and 2012, *Laricobius* adults were field collected in the vicinity of Banner Elk, North Carolina, USA. (36.165643°N, 81.872118°W). Eastern hemlock foliage infested with hemlock woolly adelgid was obtained from trees near the *Laricobius* collection sites described previously, whereas uninfested eastern hemlock foliage was collected in South Burlington, Vermont, USA (44.4669° N, 73.1714° W). All clipped foliage was similarly handled and stored until used in the bioassays.

Two behavioral bioassays, conducted in the winters of 2011 and 2012, were used to test the ambulatory responses of adult *L. nigrinus* to various stimuli in a four-chambered olfactometer (Analytical Research Systems, #OLFM-4-C-2440PE, Gainesville, Florida, USA) identical to the one described in Wallin *et al.* (2011). One of the following four stimulus treatments was assigned to each glass chamber, which in turn was attached to a randomly chosen arm of the olfactometer: a

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Table 1

Ambulatory responses of *L. nigrinus* individuals to odors from host foliage, prey and conspecifics in a four-way olfactometer over two years of bioassays. Response values sharing the same letter are not significantly different ($p>0.05$).

Stimulus Field	Proportion of Choice by Year		
	2011 (N=55)	2012 (N=31)	Pooled years (N=86)
Feeding Beetle	0.13	0.16	0.14 ^b
E. Hemlock with HWA	0.35	0.29	0.33 ^a
E. Hemlock	0.24	0.26	0.24 ^{ab}
Blank Control	0.15	0.13	0.14 ^b
Center Field	0.15	0.16	0.15 ^b
	$\chi^2=9.273$ $p=0.055$	$\chi^2=3.032$ $p=0.552$	$\chi^2=11.791$ $p=0.019^*$

A Cochran Q test was completed for each year of data and the pooled data set. An asterisk indicates a significant difference in preference ($p<0.05$).

blank control (empty chamber), eastern hemlock foliage, eastern hemlock foliage infested with hemlock woolly adelgid, and eastern hemlock foliage with *Laricobius* feeding on hemlock woolly adelgid (hereafter called the feeding-beetle treatment). In the feeding-beetle treatment, a beetle was placed on a piece of adelgid-infested foliage and allowed to settle and begin feeding prior to placement of the foliage in the chamber. Foliage was infested with at least 10 adelgids per centimetre of twig length, and pieces of foliage with approximately equal densities of adelgids were used for each treatment and replicate. The foliage containing the feeding beetle was not placed into the glass chamber until the feeding beetle was visibly nestled among the adelgid wax and no longer moving. Between each replicate, foliage and the feeding beetle were replaced, and the chambers were cleaned with ethanol and allowed to dry. Chambers were randomly reassigned for each replicate.

Responses of individual *Laricobius* to treatments were measured using methodology similar to Wallin *et al.* (2011) and Arsenault (2013). A single individual was placed in the center of the arena, equidistant from the entrance of each arm. Individuals were allowed to walk about the arena for up to 10 minutes, and their choice was recorded. Bioassays were completed, and final positions were recorded when: 1) a beetle remained in a field boundary for at least one minute; 2) the 10-minute time limit was reached; or, 3) a beetle attempted to crawl into an arm. After bioassays, *Laricobius* specimens were identified as *L. nigrinus*, native *L. rubidus* or hybrids, via microsatellites as described in Havill *et al.* (2012) at the USDA Forest Service Northern Research Station, in Hamden, CT. Only *L. nigrinus* were included in the analysis so as to not confound the data. The 2011 and 2012 bioassays included 55 and 31 *L. nigrinus* beetles, respectively. Because methodology for live sexing of *Laricobius* was not published until after the completion of this study, and *Laricobius* does not orient using pheromones (Shepherd *et al.* unpublished data), beetles were not sexed as part of this study.

The proportions of beetles choosing various treatment fields for their final positions in the olfactometer were compared using the Cochran Q test. Analyses were made separately for each year (2011; 2012), as well as for pooled data from both years.

Laricobius nigrinus responded to odors in the olfactometer by walking in the arena and choosing a stimulus field over the centre field or blank chamber in both 2011 and 2012. Treatment stimulus had significant effect on the proportion of *L. nigrinus* choosing a field in 2011, and when the 2011 and 2012 data were pooled (Table 1). In the pooled data, a greater proportion of *L. nigrinus* chose the stimulus field containing eastern hemlock infested with hemlock woolly

adelgid than the other fields (Table 1). The stimulus field containing the feeding beetle was consistently among the least preferred options (Table 1).

Laricobius nigrinus reliably responded to host foliage with hemlock woolly adelgid, but there was no significant difference in preference for infested versus uninfested foliage. As described in Wallin *et al.* (2011), the similarity in preference between these two host treatments may be due to the low detectability of the adelgid in the olfactometer without added information from visual and tactile stimuli. This phenomenon has been described as the reliability–detectability problem (Vet *et al.* 1991), where the magnitude and surface area available for release of olfactory cues is much greater for the foliage than for the prey. However, odors from hosts alone are not necessarily a reliable indication of prey availability, so predators can use a combination of these, as well as herbivore-induced volatile cues emitted by the host when foliage is wounded through feeding (Agrawal 1998; Dicke and Van Loon 2000; Havill and Raffa 2000; Radville *et al.* 2011). The data presented here support that *L. nigrinus* predators are attracted to hemlock woolly adelgid and host odors, and may use hemlock foliage as a proxy due to low detectability of prey.

In summary, this study presents evidence that *L. nigrinus* may use volatile cues to avoid feeding conspecifics when foraging for prey. Use of chemical cues may enhance dispersal of *L. nigrinus* populations across the *A. tsugae* prey resource on hemlock trees in the field. Additional research is needed to better understand how volatile cues may be integrated with visual, tactile, or other stimuli during the host-finding behavior of this important biological control agent in North American hemlock forests.

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SCIENTIFIC NOTE

First record of the *Lasioglossum (Dialictus) petrellum* species group in Canada (Hymenoptera: Halictidae)**JENNIFER HERON¹ and CORY S. SHEFFIELD²**

In the past decade, several taxonomic publications focusing on and/or including the Canadian bee fauna (e.g., Gibbs 2010, 2011; Gibbs *et al.* 2013; Sheffield *et al.* 2011; Dumesh and Sheffield 2012; Williams *et al.* 2014) have greatly increased our knowledge of species diversity in the country. In turn, this increased knowledge has facilitated the assessment of seven bee species by the Committee on the Status of Endangered Wildlife in Canada (COSEWIC) (www.cosewic.gc.ca/eng/sct1/searchform_e.cfm) and the first national general status assessment of Canada's bees to be published later this year in the Wild Species 2015 report (R. Hébert, pers. comm., 2015). However, our knowledge of the bee fauna of Canada, though comprehensive, still contains many gaps due to the expansive size of the country, diverse plant communities and habitats, and the high proportion of these habitats that have been poorly sampled for bees or have not been sampled at all.

The Western Interior Basin of southern British Columbia is the smallest ecozone in Canada (approximately 56,500 km²), and also one of the most species rich. The high diversity of bees in this ecozone in Canada is largely due to the Western Interior Basin being the northernmost extent of the Great Basin shrub–steppe grasslands and dry interior low-elevation forest ecosystems that extend from Mexico, northward through the central–western United States, to the southern in the Okanagan and Similkameen valleys (Pryce *et al.* 2006). The Okanagan and Similkameen valleys are also home to a number of angiosperms (see British Columbia Conservation Data Centre 2015) and invertebrates (see Scudder 1994; British Columbia Conservation Data Centre 2015) that, in Canada, are geographically restricted to this area, often representing the northern edges of the species' ranges in the Great Basin (Straley *et al.* 1985). Arid climate conditions, like those of this region, are known globally to promote bee diversity (Michener 1979, 2007) and thus contribute to the Western Interior Basin's high diversity of bees. The region contains half of Canada's bee species, and approximately one-third of those species occur in Canada only in the Western Interior Basin (Sheffield *et al.* 2014).

From an entomologist's perspective, the lowlands and surrounding hillsides of the Western Interior Basin may hold many more surprises. For instance, two of the 19 new species of *Lasioglossum* (subgenus *Dialictus*) described by Gibbs (2010) seem to be restricted in their distributions in Canada to this area, as are the ranges of four additional species covered in the same comprehensive 2010 treatment. Similarly, almost 25% of *Megachile* species found in Canada are found only in this ecozone (Sheffield *et al.* 2011).

The bee fauna of the Western Interior Basin may also be among Canada's most vulnerable. Large portions of the low-elevation grassland habitats are fragmented by urban, rural and agricultural development, which is known to have severe impacts on bees within the arid habitats of the Great Basin in the United States (e.g., Cane *et al.* 2006). As such, the Western Interior Basin has great conservation value to Canada, and it is important to fully document the species in the area.

The purpose of this scientific note is to record for the first time the presence of a member of the *Lasioglossum petrellum* species group in Canada (as described by Gibbs 2009), and to summarize characteristics to help entomologists recognize it among the *Lasioglossum* subgenus *Dialictus* in Canada, a group containing at least 84 other species that Gibbs revised (2010).

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The *Lasioglossum petrellum* species group is considered distinctive within the subgenus *Dialictus* in North America, based on both morphological and molecular characters (Gibbs 2009). Members of both sexes have elongate heads—particularly the malar space—compared to other *Dialictus* in North America (Fig. 1). In addition, females typically have reddish to orange–yellow coloration on the metasoma and lack an acarinarial fan of appressed hairs on the basal area of tergum 1 (Gibbs 2009); among other Canadian *Dialictus* (following the classification of Gibbs *et al.* 2013), this characteristic would be shared by only females of *L. ruidosense* (Cockerell) *s.l.* and *L. versans* (Lovell), both which have dark abdomens (Gibbs 2010). The male genitalia in the *L. petrellum* species group have a unique retrorse lobe of the genital capsule; the lobe is greatly reduced compared to most other North American *Dialictus* and is replaced by a small tubercle with bristles (Gibbs 2009; Fig. 2B).

Two male specimens, deposited in the Royal Saskatchewan Museum, Regina, Saskatchewan (RSKM_ENT_E-90049, RSKM_ENT_E-90050), were collected in the Western Interior Basin, from Mt. Kobau within the South Okanagan Grasslands Protected Area, west of Osoyoos, in southern British Columbia [49.11064, –119.66534; 1175 m; collected on Aug. 29, 2013, by C. Sheffield, J. Heron, L. Gardiner, and O. Dyer; ex. *Antennaria*]. Although the specimens from British Columbia were clearly members of the *L. petrellum* species group (as per Gibbs 2009), specific identification could not accurately be made by morphology alone, as males of two of the species are unknown.

To resolve this, tissue samples from these specimens were sequenced for the DNA barcode region of cytochrome c oxidase subunit 1 (Sheffield *et al.* 2009), and the sequences were compared to those analyzed by Gibbs (2009). DNA sequences, photographs, and specimen data are now in BOLD (Project LEPTC, BOLD Process IDs WASPS304-14, WASPS303-14), with BankIt (1853627) and GenBank accession numbers (KT695599, KT695600). The resulting sequences from material from British Columbia shared 99.2%–99.54% similarity with specimens from California (*L. tuolumnense* Gibbs) and 98.2%–98.6% similarity with specimens from Utah (*L. griswoldi* Gibbs). *Lasioglossum tuolumnense* (Fig. 2) was described from specimens collected from high elevations in the Sierra Nevada in California, and until this account of its presence in

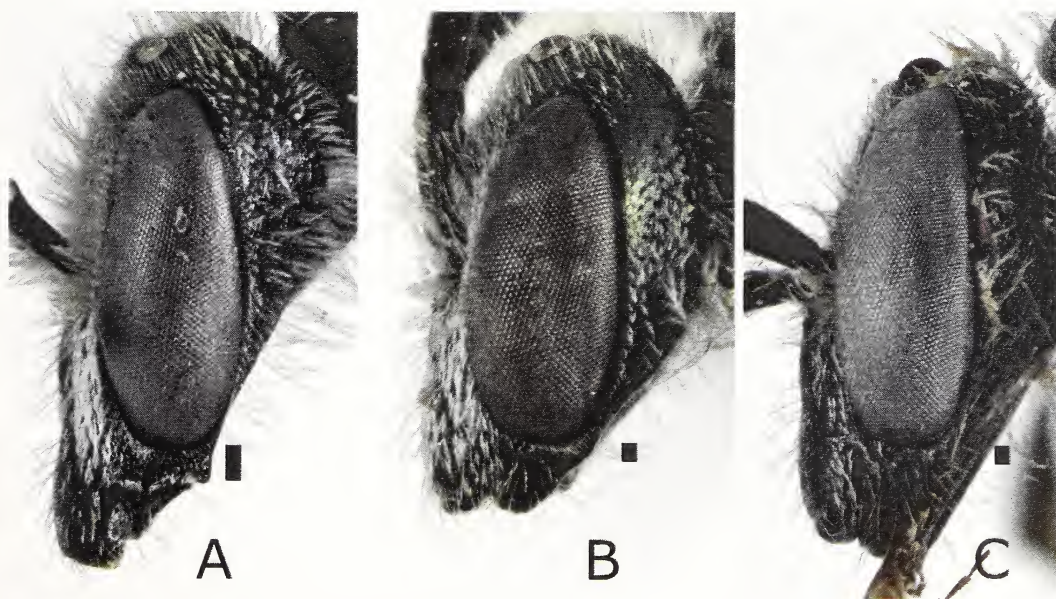


Figure 1. Lateral view of the heads of male A) *Lasioglossum (Dialictus) tuolumnense* Gibbs, B) *L. (D.) anomalum* (Robertson), and C) *L. (D.) cressonii* (Robertson); images scaled to show comparable compound eye length. Black boxes show the respective malar space height of each species.

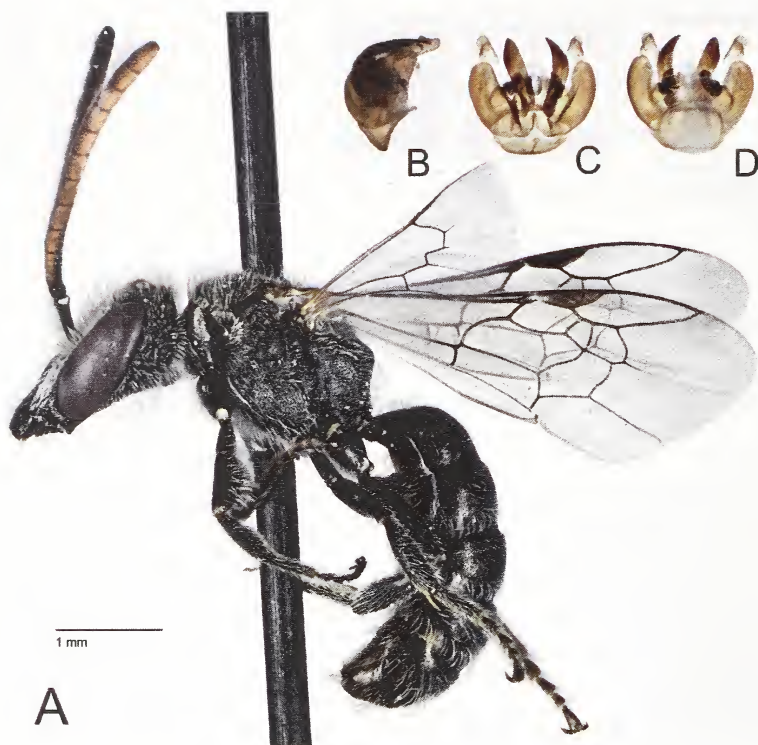


Figure 2. *Lasioglossum (Dialictus) tuolumnense* Gibbs from British Columbia, Canada. A) Male lateral view, with genitalia in B) lateral, C) dorsal, and D) ventral views.

western Canada, was known only from that state (Gibbs 2009). Thus, it is the only member of the *L. petrellum* species group known from Canada and all other species are currently known from the southwestern United States (California, Arizona, Colorado, Utah, and Texas) and adjacent Mexico (Gibbs 2009; Scott *et al.* 2011).

Gibbs (2009) addressed the close relatedness of *L. tuolumnense* and *L. griswoldi* genetically, suggesting their monophyly was not supported in all phylogenetic analyses of DNA barcode data, despite distinct morphological differences in the females. Unfortunately, the male of *L. griswoldi* is unknown (Gibbs 2009), and females were not found in the present study, so morphological comparisons of all taxa cannot be made at present. As such, because of the morphological similarity of the male specimens from British Columbia to *L. tuolumnense* and the high DNA barcode similarity, including a shared fixed-nucleotide substitution as per Gibbs (2009), we hesitate at this time to consider the Canadian representative of the *L. petrellum* group a distinct new species, despite large geographic separation (> 1000 km) from the most similar taxa in the southwestern United States. Additional sampling in high-elevation regions between the Sierra Nevada and the mountains of southern British Columbia (Okanagan and Similkameen) may resolve this species' range, although at present most species in the *L. petrellum* species group seem to be restricted geographically (Gibbs 2009).

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SCIENTIFIC NOTE

Coexistence of *Cerceris fumipennis* and *Cerceris nigrescens* colonies in Merritt, BCT. KIMOTO¹, M. BUCK², P. D. CARELESS³, and J. ROBERTS⁴

ABSTRACT

Cerceris fumipennis Say (Hymenoptera: Crabronidae) is a solitary ground-nesting wasp that provisions its subterranean nests with paralyzed jewel beetles (Coleoptera: Buprestidae). The first BC colony of *C. fumipennis* was discovered in 2012 at Central Park in Merritt. At the same location, in 2014, five female *Cerceris nigrescens* Smith were collected; one had captured a clover root weevil, *Sitona hispidulus* Fabricius (Coleoptera: Curculionidae). Single *C. nigrescens* and *C. fumipennis* nests were excavated; the former constructed brood cells from 3.8 to 7.6 cm underground, whereas the latter dug cells 8 to 15.3 cm deep.

Cerceris fumipennis Say (Hymenoptera: Crabronidae) is a solitary ground-nesting wasp that occurs primarily east of the Rockies (Scullen 1965). Female wasps dig underground nests comprised of cells containing wasp larvae; only females provision these cells with paralyzed buprestid beetles, upon which the larvae feed and develop (Hook and Evans 1991; Marshall *et al.* 2005). As these wasps are adept at finding uncommon and arboreal species, *C. fumipennis* has been used as a biosurveillance tool to detect emerald ash borer, *Agrilus planipennis* Fairmaire (Coleoptera: Buprestidae), and to discover new jurisdictional records for other species (Marshall *et al.* 2005; Rutledge *et al.* 2013).

In 2012, a *C. fumipennis* colony was discovered in compact soil next to the gravel parking lot at Central Park in Merritt, BC (Figure 1; Kimoto and Buck 2015). It had been 77 years since these wasps were identified in BC, and this record represented the first known nesting colony in the province (Kimoto and Buck 2015). The discovery of this and two other colonies in BC (Kimoto *et al.* in press) indicates the potential to utilize *C. fumipennis* as a survey detection tool for non-indigenous jewel beetles. Unlike *C. fumipennis* in eastern North America, no data currently exist regarding the prey captured by *C. fumipennis* in BC. In order to determine if BC *C. fumipennis* could be successfully used as a detection tool, data on the size and diversity of prey was collected in 2014 (Kimoto *et al.* in press). Here, we present observations of the coexistence of *C. fumipennis* colonies with those of another wasp species, *Cerceris nigrescens* Smith.

During July and August, 2014, clear plastic cups were placed over various nest entrances at the *C. fumipennis* colony in Central Park. Upon returning to her nest, a female *C. fumipennis* will circle the blocked nest entrance, and she and her prey can be readily captured with a net. On 7 August 2014, five female *Cerceris nigrescens* were netted; one grasped a clover root weevil, *Sitona hispidulus* Fabricius (Coleoptera: Curculionidae).

Cerceris nigrescens is widely distributed throughout North America. Females provision their nests with weevils including species of *Sitona*, *Hyperodes* and *Rhinusa* (= *Gymnaetron*) (Scullen 1965). The clover root weevil is indigenous to Europe; the first North American record is from New Jersey in 1875 (Campbell *et al.* 1989). It is known across Canada (except for Manitoba) (Bousquet *et al.* 2013). The larvae feed on the roots and adults feed on the foliage of clover,

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alfalfa, and vetch (Campbell *et al.* 1989). *Sitona hispidulus* is a major prey species of *C. nigrescens* in many different locations (Scullen 1965; Evans 1971). The *C. nigrescens* specimens collected from Merritt have been deposited at the Royal Alberta Museum, Edmonton, Alberta, Canada. The *Sitona hispidulus* specimen is archived at the Pacific Forestry Centre Arthropod Collection (Victoria, BC).

Cerceris fumipennis entrance holes are approximately 5–7 mm in diameter. Although the primary focus of our study was *C. fumipennis*, by August we observed smaller nest entrances among those belonging to *C. fumipennis*. The *C. nigrescens* carrying the clover root weevil was captured in a sweep net while it approached one of these smaller nest entrances.

On 7 August 2014, a *C. fumipennis* nest at least two feet from any other *C. fumipennis* nest was excavated; a hammer and chisel was used to dig a 35 x 30 x 20 cm deep hole (Figure 2). The hard, compact soil was broken into small pieces and examined for beetle prey. Upon discovering a cell containing insects, we measured its depth below the surface with a tape measure. At 3.8 cm below the surface, we found one small, black unidentifiable weevil. At 7.3 and 7.6 cm beneath the surface, many black elytra similar to those associated with the first buried weevil were uncovered. Neither the weevil nor the elytra were observed to occur within distinct cells, but as the primary focus was collecting data on *C. fumipennis*, these may have been overlooked. No cells containing buprestids were discovered down to 7.6 cm, but from 8 to 15.3 cm beneath the surface, the absence of weevils and the presence of buprestids within larger jellybean-shaped cells suggest that *C. nigrescens* and *C. fumipennis* cells are vertically segregated.

The presence of *C. nigrescens* nests with *C. fumipennis* or even other *Cerceris* species is not unique. In southern Ontario, eleven other *Cerceris* species were observed nesting at nine *C. fumipennis* colonies, but *C. nigrescens* nests were only found at one site (unpublished data). Similar to *C. fumipennis*, *C. californica* Cresson also preys upon buprestid beetles, but is restricted to western North America from Texas through New Mexico, Arizona, California, and northward into BC (Scullen 1965). *Cerceris californica* colonies were recently discovered in Washington State, and various wasps and bees occurred at these colonies (Looney *et al.* 2014). *Cerceris nigrescens* was recorded at a *C. californica* colony in Yakima, WA, but the presence of



Figure 1. *Cerceris fumipennis* and *C. nigrescens* colonies (red circle), Merritt, BC (50.11875°N, 120.78348°W).



Figure 2. Excavation of *Cerceris fumipennis* and *C. nigrescens* nests, Merritt, BC.

C. nigrescens nests is unconfirmed, because adults were not observed flying toward burrows (C. Looney, Washington State Department of Agriculture, personal communication).

The *C. nigrescens* and *C. fumipennis* colonies in Merritt represent the first records of these two species nesting together in BC. The presence of *C. nigrescens* should not affect the use of *C. fumipennis* as a bio-surveillance tool, because the former strictly provisions their nests with weevils. In fact, the occurrence of these two *Cerceris* species at a given site would allow an observer to monitor the area for both jewel beetles and weevils.

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OBITUARY



Philip Arthur Jones
(1 March 1924 – 8 January 2015)

Phil Jones, entomologist, naturalist, and long-time member of the Entomological Society of BC, was born in Prince George, BC on 1 March 1924. He died in Vernon, BC on 8 January 2015, two months short of his 91st birthday.

Although he spent his early years in Prince George, Phil moved to Smithers in 1937 and, after completing high school, and fascinated by the natural world, he enrolled in Agriculture in the winter session of 1942-43 at the University of British Columbia. World War II intervened, however, and, from 1944 to 1945, Phil served in the Royal Canadian Navy Volunteer Reserve-Signals Branch. Returning to UBC in 1946, he graduated in 1949 with a BSA, majoring in horticulture. During his undergraduate years, Phil spent the summers of 1947 and 1948 as a student assistant in forest entomology with the Canada Department of Agriculture in Vernon, BC and spent time at the Trinity Valley Field Station, a place guaranteed to turn any young naturalist into an entomologist. After graduation from UBC, Phil was promoted to Assistant Forest Biologist and continued to work in the Vernon office until September 1952.

Phil then left BC for the University of Wisconsin in Madison, where he earned his MSc in Entomology in 1956. His doctoral studies in forest entomology at Wisconsin were interrupted from July 1958 to July 1960, when he was hired as a Research Officer with the Canada Department of Agriculture, Forest Entomology, in Ottawa. He returned to the University of Wisconsin in 1960 to study biological control of forest insect pests and, in 1963 he received his PhD in Entomology with a minor in Plant Ecology.

In 1964 Phil accepted a position as Assistant Professor in the Entomology-Zoology Department at South Dakota State University in Brookings. He combined his duties as State Survey Entomologist with field research projects on the control of agricultural insect pests in small grain, row and forage crops throughout South Dakota. He edited, and contributed to, the

weekly South Dakota Insect Survey Newsletter, which was distributed to extension workers, growers and producers in South Dakota and adjoining states. He also provided South Dakota information to US Department of Agriculture surveys of insects of economic importance in the north-central states.

Phil moved to Burlington, Ontario in 1974 as Technical Director for Niagara Chemicals and, in 1977, he joined the Environmental Protection Service (Commercial Chemicals Branch), Environment Canada, in Ottawa as a Senior Scientist. His first assignment was a major technical review of chlorophenols in the environment. Other environmental reviews followed, including those for the commercial chemicals derived from organotins and chlorobenzenes.

Phil transferred to the Vancouver office of the Environmental Protection Service in June 1992 as a Senior Pest Management Specialist. He retired to Vernon in November 1993, returning to the Okanagan Valley that he fell in love with 45 years earlier as a young university student.

Throughout his long career Phil was an author of many academic papers and publications in forest and agricultural entomology. But he was interested in many things, from history (a recent project was the documentation of the history of the Bulkley Valley, where he spent part of his youth) to photography, from skiing to nature education. What I saw mostly, though, in my friendship with Phil, was a love of natural history and a deep interest in people. He frequently phoned me at the Royal BC Museum for information, or sent me specimens and insect photos for confirmation of identification. Often these requests were passed along from friends or other Vernon naturalists; Phil was a great facilitator. During his first years as a professional entomologist, Phil was one of the people connected to the Forest Insect Laboratory in Vernon who, in 1951, formed the North Okanagan Naturalists' Club. He served as its first secretary-treasurer in 1951- 1952. This organization became a popular and powerful force in BC nature study and conservation and, in May 2014, for his significant contributions, Phil was elected a life member.

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Symposium Abstracts: Outbreaks, Climate Change, and Shifting Priorities: The Future of Forest Entomology

Entomological Society of British Columbia Annual General Meeting,

Henry Grube Educational Centre in Kamloops, BC, October 16-17, 2015

Forest Entomology in the BCFLNR

Tim Ebata, BC Ministry of Forests, Lands and Natural Resource Operations, Victoria, B.C.

Forest entomologists have been hired by the province since the late 1970s and have played a vital role in applying their skills and expert advice to help manage forest insect pests in British Columbia. This presentation describes the current roles and responsibilities of professional forest entomologists in the B.C. public service and provides insight into how important these positions will be in the future—Some challenges facing the maintenance and recruitment of these positions are also discussed.

Dryocoetes confusus, shaping subalpine fir forests

Lorraine MacLauchlan, Ministry of Forests, Lands and Natural Resource Operations, Thompson–Okanagan Region, Kamloops, B.C.

The western balsam bark beetle is considered the most destructive mortality agent in subalpine-fir ecosystems, yet the actual impact of this beetle is not well quantified. Results from two long-term studies highlight the significant and ongoing impact by western balsam bark beetle in high-elevation forests of southern British Columbia. The wet, cold Engelmann Spruce–Subalpine Fir ecosystem (ESSFwc) is the most predominant ecosystem in southern B.C. that contains subalpine fir suffering moderate levels of attack by western balsam bark beetle—on average 0.8% trees killed annually. The very dry, cold Engelmann Spruce–Subalpine Fir (ESSFxc) ecosystem sustained the highest levels of mortality: averaging 1.6% trees killed annually (47% average in-stand mortality by 2014). Therefore, it is reasonable to conclude that, with continued and increasing climatic stresses in the ESSFwc, this ecosystem will likely experience higher and more severe levels of attack in the coming decades. The preponderance of red attack in the 2014 assessment compared to the first assessment in 1997 suggests that mortality is occurring at higher rates now than two decades ago.

Predicting Outbreaks of the Western Spruce Budworm: Are we there yet?

Brian Van Hezewijk, Natural Resources Canada, Victoria, B.C.

The western spruce budworm, *Choristoneura occidentalis* Freeman, is the most important defoliator of spruce–fir forests in western North America. If we want to better predict forest carbon balances and regional timber supplies, or gain a better understanding of climate change feedbacks, I argue that we need to have an accurate and usable population model for this species. In this presentation, I describe some of the past research that has shaped our understanding of western spruce budworm dynamics, our current work aimed at filling important gaps in our knowledge—particularly for low-density populations—and describe a new spatially explicit model intended to both describe and project populations at the regional scale.

Western Spruce Budworm in a Changing World

Lisa Poirier, University of Northern British Columbia, Prince George, B.C.

In recent years, western spruce budworm, among other forest insects, has reached outbreak levels in areas where it had not been recorded historically. The influence of climate on insect outbreaks is clear and well documented. The complex and changing ecological interactions among the abiotic and biotic environment, the host, and the insect are still challenging to unravel. For example, in areas with shorter growing seasons, the ability of the host tree to compensate for defoliation by production of late buds may be inhibited, possibly increasing the risk of mortality.

Furthermore, interactions between insects can affect outcomes. Forest managers often suggest that defoliation of Douglas-fir by insects such as western spruce budworm can facilitate later attack by Douglas-fir beetle. Initial spatiotemporal analysis has suggested that, on a landscape scale at least, beetle attack may actually be less likely following a western spruce budworm outbreak. Our efforts to predict the future, even in an era of increasingly rapid change, can often be meaningfully informed by looking to the past. Ongoing dendrochronological analyses examine western spruce budworm and Douglas-fir beetle history near the northern limits of interior Douglas-fir forest type. These reconstructions may help to identify the abiotic and biotic stresses that could trigger or intensify insect outbreaks. Both current and historical data about the world inhabited by these insects are needed to make reliable predictions about their future impacts.

Evolution of Decision Support for Forest Ecosystem Management: Towards Open Modelling and Data

Bill Riel and Alex M. Chubaty, Pacific Forestry Research Centre, Victoria, B.C.

Forest ecosystem decision support systems evolved out of a need to apply the results of scientific research to forest management and policy. Initially, these were closed systems, limited by technologies and data availability at the time. The recent availability of large spatial datasets and high-performance computing has enabled creation of new systems-modelling approaches. We examine decision support tools and models used for mountain pine beetle management in B.C. by discussing the advantages and limitations of these historical approaches. Current development of open and scalable modelling platforms seeks to overcome many of the historical limitations and provide cross-disciplinary integration, along with enhanced transparency, accountability, and scientific reproducibility.

Presentation Abstracts

Entomological Society of British Columbia

Annual General Meeting,

Henry Grube Educational Centre in Kamloops, BC, October 16-17, 2015

Current insect pest issues in the Southern Interior of British Columbia

Susanna Acheampong, British Columbia Ministry of Agriculture, Kelowna, B.C.

Insect pests of concern in field and horticultural crops are discussed. Pest species include grasshoppers and flatheaded borers.

The spatial and temporal distribution of total drosophilids in a cherry orchard

Amanda Chamberlain, Department of Biology, University of British Columbia–Okanagan, Kelowna, B.C.

This study characterized the distribution of total drosophilids within a cherry block and tree canopy by sampling ca. 1,700 cherries over six weeks in 2015. Our findings promote a better understanding of the spatial and temporal dynamics of drosophilids, including *Drosophila suzukii*, and should be considered in future management programs.

Flipping Phenotype: Creating resistant *Aedes aegypti* to prevent dengue transmission

Heather Coatsworth, Paola Caicedo, Clara Ocampo, and Carl Lowenberger, Department of Biological Sciences, Simon Fraser University, Burnaby, B.C.

Dengue is the most widespread arbovirus, transmitted by *Aedes aegypti*. In Colombia, 30 percent of feral *Ae. aegypti* are resistant to dengue. We used RNA sequencing to compare gene expression in susceptible and resistant mosquitoes. We knocked down gene expression of proviral genes and reversed the phenotype from susceptible to resistant.

Preliminary lifetable analysis on diamondback moth (*Plutella xylostella*) in British Columbia

Tina Dancau^{1,2}, Dave Gillespie¹, Peggy Clarke¹, and Eleanor Stewart^{1,3}: ¹Agriculture & Agri-Food Canada, Summerland, B.C.; ²Department of Biological Sciences, Simon Fraser University, Burnaby, B.C.; ³Department of Biology, University of Victoria, Victoria, B.C.

The diamondback moth (*Plutella xylostella*) is a globally distributed pest that infests *brassicaceae*, such as canola, mustards and cabbage. We looked at mortality factors affecting the diamondback moth in B.C. through a preliminary lifetable analysis. A parasitoid assemblage and implications for future management are also discussed.

Development consequences for the diamondback moth (*Plutella xylostella*) in a warming world

Dave Gillespie¹, Tina Dancau^{1,2}, Eleanor Stewart^{1,3}, and Peggy Clarke¹: ¹Agriculture & Agri-Food Canada, Summerland, B.C.; ²Department of Biological Sciences, Simon Fraser University, Burnaby, B.C.; ³Department of Biology, University of Victoria, Victoria, B.C.

Degree-day models are important tools in insect biology, but are mostly developed from constant temperature data. We ask if degree-day models based on constant temperature conditions can predict the performance of insects under variable temperatures. We used the diamondback moth as a research model.

The European fire ant: an update on a growing issue

Rob Higgins, Department of Biology, Thomson Rivers University, Kamloops, B.C.

Since the initial finding of the European fire ant in North Vancouver in 2010, it is now known that the ant is established in almost every municipality in the Metro Vancouver area, in some parts of the Fraser Valley, and on Vancouver Island. The cryptic nature of the nests and polygynous structure of the colonies are two of several attributes that make control a challenge. I present an

update on the distribution of this ant, discuss the impacts of this ant, and provide some initial results at control.

Sexual communication in *Xenos peckii* (Strepsiptera: Xenidae), a parasitoid of the paper wasp *Polistes fuscatus*

Michael Hrabar, Huimin Zhai, Regine Gries, Robert Britton, Paul Schaefer, and Gerhard Gries, Department of Biological Sciences, Simon Fraser University, Burnaby, B.C.

Strepsipterans are peculiar, obligate entomophagous endoparasitoids. The free-living, winged adult males live only for a few hours, in which time they must locate the neotenic, host-bound, females. We identified, synthesized, and field-tested the sex pheromone of *Xenos peckii*, a parasitoid of North American paper wasps.

Balsam woolly adelgid, *Adelges piceae*: slow but deadly

Tracy Hueppelsheuser, British Columbia Ministry of Agriculture, Abbotsford, B.C.

Balsam woolly adelgid (BWA) has been present in North America for more than 100 years, spreading from east to west. It has killed forests of true fir (*Abies* spp.) trees across the continent. British Columbia's interior has been one of the last BWA-free areas. However, there is evidence of incursion. In order to gain a better understanding of the insect's current distribution in B.C., I encourage all practitioners to watch for this pest in interior zones, and report suspicious finds. I share biology, impacts, and what to look for, as well as discuss implications of establishment of this pest.

Population structure in *Diplolepis variabilis*: is *Wolbachia* present at low frequencies?

R.G. Lalonde, D. Bartkiewicz, J. Bannerman, Department of Biology, University of British Columbia–Okanagan, Kelowna, B.C.

The commonest gall on *Rosa woodsii* in the Okanagan is formed by *Diplolepis variabilis*. Prior assaying for the presence of *Wolbachia* in this species failed to find the symbiont. Our earlier work showed some sites have high frequencies of female emergents. Accordingly, we screened samples taken along a north–south gradient and found partial infestation by *Wolbachia* at some sites.

How climate and host behaviour influence nucleopolyhedrovirus infection dynamics in the western tent caterpillar

Paul R MacDonald¹, Judith H Myers², Jenny S Cory¹: ¹Department of Biological Sciences, Simon Fraser University, Burnaby, B.C.; ²Department of Zoology, University of British Columbia, Vancouver, B.C.

Western tent caterpillar (WTC) populations display cyclical dynamics with 8- to 11-year periodicity in southwestern B.C. Long-term population data show that nucleopolyhedrovirus (NPV) is an integral component of these dynamics, with high incidence of viral mortality coinciding with substantial population decline. Our research involves two areas of NPV transmission that remain unclear. Firstly, how climate influences NPV transmission, and; secondly, how host behaviour contributes to transmission and the eventual formation of an epizootic. Using historical population and weather data, we have shown a correlation between warm springtime temperatures and heightened levels of NPV infection. We conducted a series of laboratory and field-based experiments that suggest possible mechanisms for this observation. Additionally, we conducted experiments that reveal whether infected hosts differ from uninfected hosts in their behaviour, and discuss how these differences could facilitate NPV transmission at the population scale. We conclude by discussing how empirical evidence and theoretical modelling may be used to predict how WTC population dynamics behave under various climate and behavioural scenarios.

Do multi-modal foraging cues attract mosquitoes (Diptera: Culicidae) to flowers?

D. Peach, R. Gries, H. Zhai, and G. Gries, Department of Biological Sciences, Simon Fraser University, Burnaby, B.C.

Mosquitoes of both sexes feed on plant sugars, including from floral and extra-floral nectaries. However, the cues they use to locate these flowers are not well understood. Attractive floral volatiles were identified, and their interaction with other multi-modal cues was investigated.

Trypanosomatid parasites of *Drosophila*

Steve Perlman and Finn Hamilton, *Department of Biology, University of Victoria, Victoria, B.C.*

Trypanosomatids are ubiquitous and diverse eukaryotic parasites of arthropods, and include the causal agents of human diseases, such as African sleeping sickness and leishmaniasis, that are vectored by blood-feeding arthropods. Yet very little is known about trypanosomatids that are not associated with blood feeding. It has long been known that *Drosophila* harbour diverse trypanosomatids, but these are poorly studied, which is especially surprising since *Drosophila* are major animal models of host–parasite interactions and animal immunity. We present our recent work characterizing *Jaenimonas drosophilae*, a newly described lineage of *Drosophila*-parasitic trypanosomatids, including experiments aimed at understanding its host range, transmission and infection dynamics, and effects on host fitness.

Host-tree selection by adult spruce budworm

Ward Strong, *British Columbia Ministry of Forests, Victoria, B.C.*

Western spruce budworm, *Choristoneura occidentalis*, is a major pest of Douglas-fir (*Pseudotsuga menziesii*) in the interior of British Columbia. Deploying budworm-resistant trees on a landscape scale would be a desirable means of limiting damage. No-choice larval performance trials on 83 known Douglas-fir genotypes indicated a wide range of host suitability, with defoliation ranging from 0% to more than 90%, suggesting a strong possibility of selecting trees resistant to larval damage. Resistance to adult host selection may also be a possibility; avoiding oviposition in the first place could be an effective management tool. A screening program for adult host preference of the same 83 tree genotypes was established. Two years of trials using insects from a laboratory colony on cuttings from the parent trees in a greenhouse were unsuccessful. For future trials we will use grafts of the parent trees, growing in soil with a screen house constructed around them, and will collect wild pupae for release inside the screen house.

Seasonal dynamics of spotted wing *Drosophila*, *Drosophila suzukii* (Diptera: Drosophilidae), on cherries and non-crop plants in British Columbia interior valleys, 2010–2014

Howard Thistlewood, *Pacific Agri-Food Research Centre, Agriculture and Agri-Food Canada, Summerland, B.C.*

D. suzukii was monitored in and around fruit orchards and in fruit from non-commercial plants nearby. A pattern of succession in host plants is related to recorded population dynamics, and to new analyses of landscapes and weather data, for understanding its overwintering, ecology, and crop damage, in the Columbia–Okanagan basins.

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